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ABSTRACT

Focusing upon a working knowledge of ecological principles as a requisite for today's society, this teacher's guide suggests numerous field studies which make pertinent use of these principles. It is designed to serve as an aid in planning student-centered activities which allow for understanding and improving the ecosystem in which they are an integral part. To assist the teacher with field activities, a series of descriptions of Brevard County, Florida, plant and animal communities is provided. The major section of the guide suggests field investigations in several areas: biomes and ecosystems, population and communities, nutrition web, aquatic ecology, and man vs. nature. Background information, purpose of the activity, materials required, and procedures to follow are enumerated with diagrams and charts drawn when necessary. Also included are ideas for water and sewage analysis, a listing of possible case studies relevant to ecological problems in Florida, and a review of procedures in selecting and developing study sites for an ecology improvement project. Miscellaneous teacher reference and resource material is appended. This work was prepared under an ESEA Title III contract. (BL)

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TEACHERS CURRICULUM GUIDE FOR FIELD ECOLOGY

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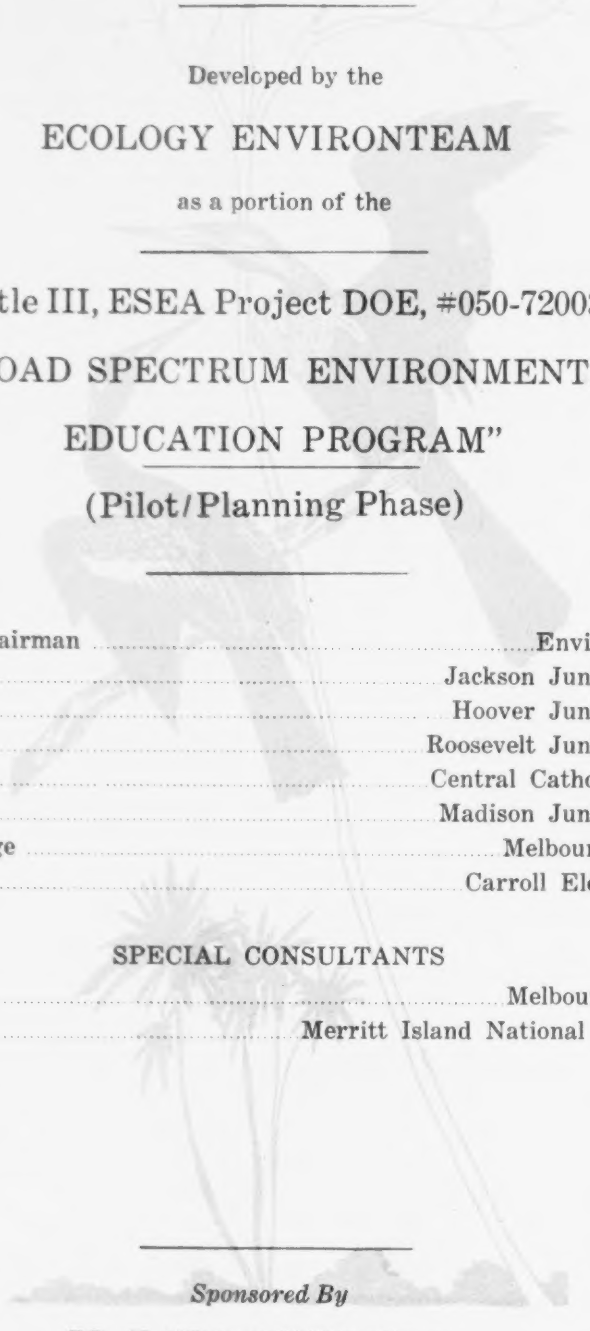
(Pilot/Planning Phase)



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PREFACE

The requirement for a broad environmental education in today's society is generally recognized; however, it is the intention here to limit the educational objectives to the teaching of ecological relationships. The specific objective of such a course of study would focus upon a working knowledge of ecological principles. This can best be accomplished through field studies which make pertinent use of these principles. There is an important need to remove the student from the confines of his school and use the local community as his classroom. Not only does the student seem to have a psychological need for truly meaningful, constructive experiences; recently developed evidence indicates that there is little transfer of "learning" from the conventional classroom to social behaviors outside of the school. This Guide will aid in planning activities needed to provide "Real World" ecological experiences for students through field studies in the natural social context of his community.

In order to meet these objectives, emphasis should be placed upon individual student and group inquiry. The ecology course should revolve around student centered activities aimed at understanding and improving the eco-system of which they are an integral part.

Certain portions of the Biological Sciences Curriculum Study (BSCS) Green Version text have been recommended in order to provide a conceptual background for laboratory, field, and community activities. Only eight of the 20 chapters in the Green Version have been recommended, since additional textbook work would interfere with the quality and quantity of laboratory and field experiences. It is felt that the eight recommended chapters will contribute to a good basic course when coupled with the materials contained in this guide and certain classroom references. Those references recommended and cross-referenced with the Green Version include the McGraw Hill book, Life of the Marsh and Life of the Pond; the Golden Press book, The Seashore; Holt, Reinhart & Winston's Ecology; and Time, Inc. Ecology.

To assist the teacher in the audio-visual aspects of the instructional program a list of films in the film library applicable to ecology has been included. Also included is a list of new audio-visual materials that have been recommended for purchase and will be earmarked for ecology.

To assist the teacher with field activities, a series of descriptions of Brevard County plant and animal communities have been provided. These are necessary for the proper interpretation of our landscape.

The present guide has drawn heavily from The Teachers Curriculum Guide for Field Ecology which was prepared as special teaching material for the pilot program developed for use in the 1971-72 school year. However, a complete revision of these materials has been accomplished; the section on Biotic Communities in Brevard County has many new additions, among which are methods for investigation of food webs, soil bacteria, algal populations, mosquito ecology, artifical ponds, microcosms, and local ocean communities.

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This entire Curriculum Guide is printed on 100% recycled paper.

PHILOSOPHY

There are many biology courses available to the secondary student today; however, none offer complete attention to the field of ecology. Instead, ecology is inserted as a chapter in the course. It is our intent to offer to the secondary teacher, a course of study through which the teacher will accomplish the following objectives:

1. Offer an opportunity for the student to exercise his innate curiosity and imagination in applying them to living systems.
2. Permit the student to become involved as a participant in the discipline, rather than an observer who is to be entertained by the teacher in a teacher-oriented "learning situation."
3. Encourage individual effort and solicit new creative approaches to concomitant problems.
4. Further encourage the student to solve problems by means of objective, yet critical, thinking as opposed to reacting emotionally to the problem.
5. Maintain the integrity of this course by establishing lines of communication with and between the participating teachers, thereby keeping the pulse of attitudes and ensuring that the objectives and philosophy of the course adhere to the guidelines along which it was written.
6. The environment should be the vehicle through which learning is accomplished.

7. It will make available to the student "real world" environmental problems that can be explored in his own surrounding.
8. Develop attitudes which result in the acceptance of responsibilities to wisely manage the environment.

REPRODUCTION OF THIS MATERIAL

Contrary to usual practice, no restrictions are placed on the use, reproduction or quotation from this Curriculum Guide if the goal is intended to improve the environmental awareness and understanding of teachers, students, and the public in general.

RECORDING INVESTIGATION DATA

There are two types of investigations; one concerns an area while the other an exercise involving one or more principles and/or methods. Therefore it is necessary to arrange the notebook using the format below when collecting data and writing results.

The investigations are the following types:

1. Investigations performed over a long period of time with unknown results until data is collected.
2. Field investigations at a school or home site.
3. Investigations in which a series of studies must be made before results can be observed.
4. Project type investigations done in the field or through research.
5. Laboratory methods to be used in field investigations.

The exercise investigations are those performed in the classroom laboratory which demonstrate a principle or method.

Before proceeding with the investigation, the following should be observed:

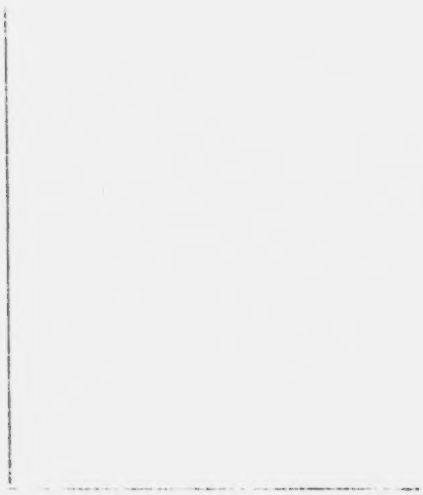
1. Read through the entire investigation before doing anything.
2. Wait for any special instructions by teacher.
3. Keep data sheets and notebooks up to date and orderly.
4. All investigations should show tabular or graphic presentation of the data with a legend and units of measure.
5. The laboratory work area and equipment shall be clean and neat at all times. Plan clean-up time during each lab.

The format of an investigation is necessary in order that the work follows a logical sequence in the performance of the investigation thereby enabling the investigator to reach a logical conclusion from data collected. Below will be found the general format to be followed in recording the investigations.

1. INVESTIGATION
2. BACKGROUND
3. PURPOSE
4. MATERIALS
5. PROCEDURE: As written in the investigation.
6. DATA: All observations and readings recorded in an orderly procedure on the data sheet with the date and any calculations and any necessary drawings.
7. QUESTIONS: Discussion questions pertaining to the interpretation and analysis of data.
8. CONCLUSION: That which was learned from this investigation.

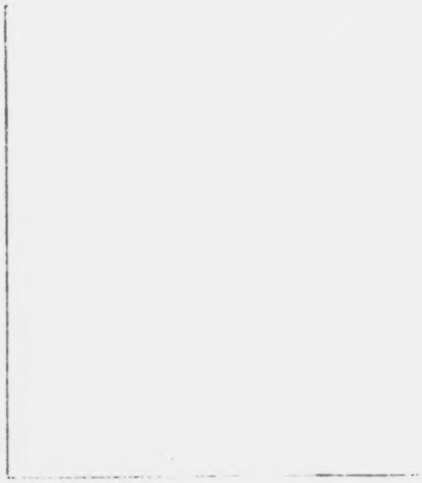
GRAPH SHEET

Below is the format for laboratory graphs. Title the graph in the space provided. Label the dependent and independent variables. Then carefully plot your graph on graph paper.



Graph A: ________________

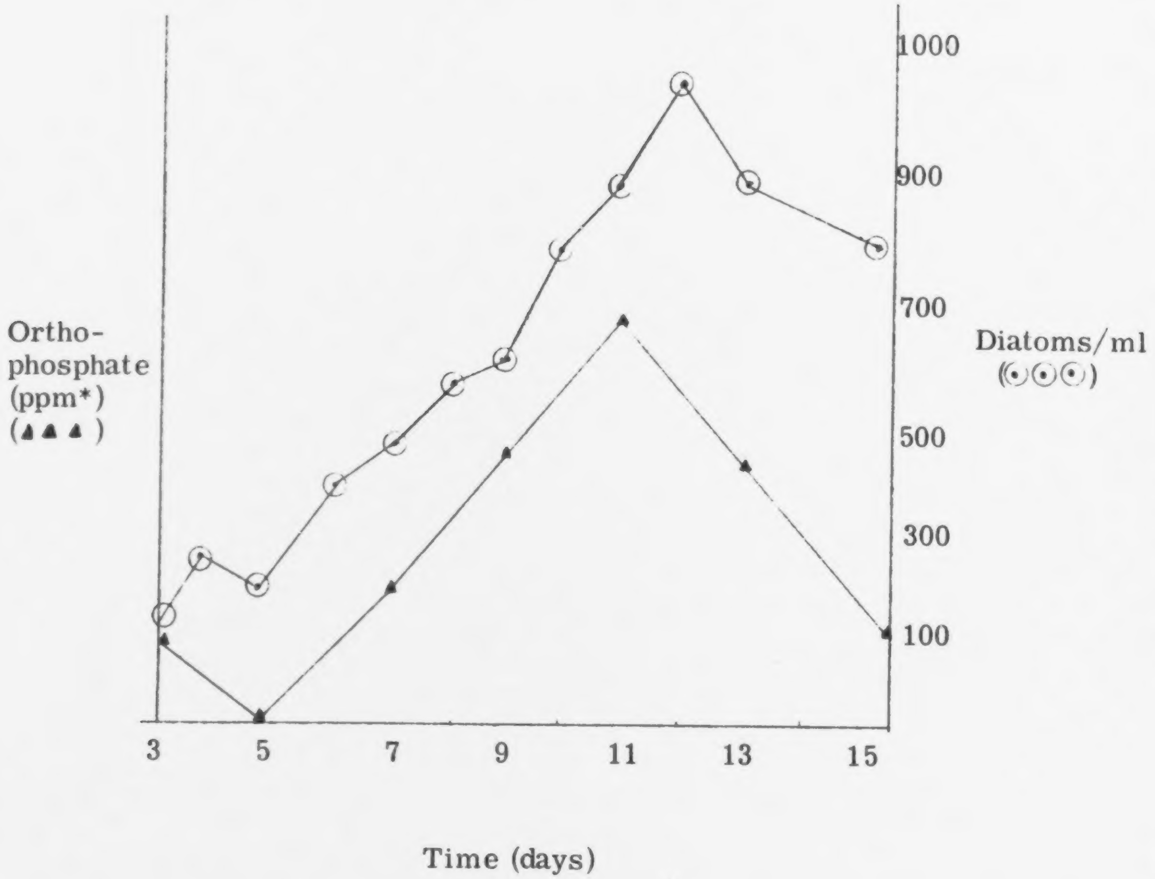
Graph B: ________________



Graph C: ________________

Graph D: ________________

EXAMPLE OF GRAPH



Graph A: A comparison of the effect of inorganic phosphate on the growth of a population of diatoms in the West Park Canal. Two inches of rain fell on October 5.

*1 ppm 1 mg/1 liter or 1 ug/1 ml

Unit of Study	Text Readings	BSCS Investigations	Curriculum Guide
1. Introduction		1:1; 1:3; 1:4	
2. Nutrition Web	Chapter I p. 2-35	1:5	
3. Community	Chapter III p. 74-94 Disease p. 216-233	7:2; 7:1; 3:1; 8:3	
4. Ecosystem	Chapter III p. 94-101 Chapter 7 p. 234-249	7:4; 7:3; 3:2	
5. Population	Chapter 2 p. 36-71 Chapter 20 p. 748-752	2:2	
6. Biomes	Chapter 8	8:2	
7. Succession a. Terrestial b. Aquatic	Chapter 8 p. 293-301 Chapter 3 p. 92-95	8:3; 9:1	
8. Hydro Ecology a. Marine b. Aquatic	Chapter 9	9:2	
9. Man vs. Nature (Pollution)	Chapter 20 p. 741-773		
10. Behavior	Chapter 15 p. 535-575	15:3	
11. Evolution	Chapter 18 p. 673-713	18:1	
12. APPENDIXES			

GEOLOGIC TIME:
Making it Comprehensible

The earth is estimated to be between 4 1/2 and 5 billion years old. If we were to equate that age with the Biblical six days of creation, we would be working with an equation in which each one second of time during that week would be the equivalent of approximately 8,000 years of geologic evolution.

On this new foreshortened scale it would take from midnight Sunday, when the Earth began to spin through space, until Tuesday noon for the globe, continents, and oceans to form. It would take until 2:00 a. m. Thursday before the simplest algae and fungi could be found, and not until 6:30 Saturday morning would advanced plants begin to proliferate.

By 5:00 p. m. Saturday afternoon things would have progressed sufficiently to allow the opening stages of the Age of Reptiles. The Great Dinosaurs would reach a peak and be off-stage completely 4 hours and 55 minutes later at 9:55 p. m. During that time the first redwoods would begin the struggle for existence in an area that ultimately would be known as Kentucky. Just before the great reptiles disappeared, our ancestors, the earliest primates, could be located hiding in the bushes, trying to avoid being stepped or fallen upon.

But, it isn't until 3 minutes and 20 seconds before midnight that Stanley Kubrick's ape in "2001" picks up a bone and invents tools. The pace accelerates drastically 3 minutes and 15 1/2 seconds later, or 4 1/2 seconds to Saturday midnight when Cro-Magnon man walks on stage, discovers weapons, and walks right off-stage again, 3 1/2 seconds later.

At 1/4 of 1 second before Saturday midnight a 33 year old bearded radical gets himself crucified for inventing the Lord's Prayer and other similar "indiscretions"; at 1/40th of one second to midnight the Declaration of Independence is written; at 1/60th of one second before midnight the industrial-technological revolution moves into high gear; at 1/80th of a second to midnight all of the inventions that made modern architecture possible are designed: steel, elevators, plumbing cores, central heating, electricity, light bulbs, telephones, automobiles.

At 1/320th of a second before Saturday midnight the atom is split over Hiroshima, and at about the same time the great DDT experiment begins. If, as some of the ecological doomsayers predict, man on earth has but 25-30 years to go unless drastic changes are made, we will be finished in another 1/320th of a second, faster than the blink of an eye. If such is to be the case, man's total duration on this planet would amount to perhaps 10 minutes. Even those stupid beasts, the dinosaurs which we accuse of blundering themselves out of existence, took 5 hours doing it.

Adapted from a talk by
David Brower of Friends of
the Earth

INTRODUCTION

BIOTIC COMMUNITIES IN BREVARD COUNTY

PINE FLATWOODS

These are open pine woodlands dominated by a single species of tree, the slash-pine. These forests often extend over vast, very level areas in pure stands. The characteristic shrubs are saw-palmetto, fetter bush, and gallberry. These are all hardy evergreens that are not killed by frost. The soil is sandy and at a depth of 2-4 feet is underlaid by an exceedingly impermeable "hard pan"; consequently, the ground water is not available to the surface soil and in times of prolonged drought the flatwoods are very arid. In wet weather, however, rainwater may remain perched at the surface for long periods of time. This water is usually contained in shallow grassy ponds which are ringed by dense stands of saw-palmetto. These ponds usually contain water in the rainy season (June-October) and are otherwise dry (November-May). The plants in this community are well adapted to fire which is the prime factor in molding the structure of this community. The special adaptations to fire are underground stems (rhizomes) as seen in saw palmetto and runner oaks, fire resistant bark and in palmetto the lack of sub-cortical cambium.

Succession in this community is usually toward a hammock-like structure, but progress in this direction is almost invariably arrested by fire which effectively eliminates most of the typically hammock species, such as oaks, or maples. In State and National Parks, carefully protected from fire, succession toward hammock species is evident.

Characteristic animals are the miniature "oak toad", the diamond-backed rattlesnake, the opposum, quail, squirrel tree frog, box turtle, red bellied wood-pecker and race-runner lizard. The burrows of the gopher tortoise are prevalent in well drained areas.

This community undoubtedly covers the largest expanse of Brevard County's terrestrial ecosystem.

HAMMOCKS

Hammocks in Florida are composed of broadleafed hardwood trees such as oaks and maples in addition to cabbage palms. Pines are conspicuous because of their variety. Hammocks often form "islands" of hardwoods in vast stands of pine (flatwoods) or form similar islands "in marshes". The soil is usually well drained, moist and has a rich humus layer. This community is the climax of the various lines of plant succession.

The vertical structure of the hammock has three layers. The characteristic canopy trees in Brevard hammocks are live oak, laurel oak, Florida maple, black gum and cabbage palm. Small trees often found in the intermediate zone include, marbleberry, the smooth barked stopper, basswood, and redbay. Species on the forest floor include wild coffee, jack-in-the-pulpit, many species of ferns and Habanela orchids. The smaller trees and shrubs are often of West Indian origin and therefore subtropical species. They are protected from frost by the canopy of larger trees of temperate origin.

FRESH WATER MARSHES

Fresh water marshes once covered a major portion of Brevard County running in a north-south direction paralleling and forming the flood plain of our largest true river, the St. Johns River. Presently, much of this once great marshland has been diked and subsequently drained by pumping the captured water over the earthen dike into the river channel. The drained land is used primarily for cattle grazing. Here the beautiful insect eating cattle egret has replaced the native fish eating species, the American and snowy egret.

The marshes of the St. John's headwaters, like those of the Everglades, often consist almost entirely of saw-grass. Vast areas of bonnets, water-lilies, pickerel weed, or arrowhead are common. Shrubs include buttonbush with round white flowers and the hibiscus-like marsh mallow. Common birds are the Anhiga, Louisiana herons, Florida mallards, and bitterns. Characteristic amphibia are the large eel-like Amphiuma, the Southern bull-frog and the green tree frog. Common reptiles are the king snake, banded, and green water snakes, ribbon snake, Nelson's turtle and the soft-shell turtle. Mammals include marsh rabbits, oppossums, and round tailed muskrats.

SAND PINE SCRUB

This type community is found on some of the highest elevations in this county; it is essentially an old sand dune association. During Pleistocene time interglacial periods found most of Brevard submerged under the sea. Melting glaciers left only a few islands, a chain of wind swept dunes running in a north-south direction. The soil is always an almost pure white sand and in some places over 40 feet in depth. The sand pine (Pinus clausa), a short needled pine with small cones often used for a Christmas tree in Florida is the dominant tree. The undergrowth consists of woody shrubs and dwarf trees which include twin oak, myrtle oak, rosemary (with small needle-like leaves) and saw-palmetto. Ground lichens (reindeer moss) are often common.

A number of species of animals seem to be confined to this habitat. Found only in the "scrub" are 4 species of grasshoppers, a spider (Lycosa ceratiola) which builds tube like holes in the sand, a bird, scrub jay, and a fence lizard (Sceloporus woodi). These species probably evolved while isolated on those Pleistocene islands which now survive as patches of "scrub." Like many other insular species, the Florida jay shows little fear of humans. These wild birds are easily trained to eat from one's hand.

The largest sand pine scrub in Florida is preserved in the Ocala National Forest and it is known as the "Big Scrub." In addition to the animal species already listed here, the "Big Scrub" has deer, black bear and wildcats.

SALT MARSHES

This plant and animal association is typically grassy with few, if any, trees or shrubs. These grassy areas form a coastal margin between the land and sea and are washed alternately by nutrient-rich runoff from the land and trace-element rich tidal waters. This habitat is, perhaps, one of the most productive of animal life, since an abundant growth of plant life and algae contribute the base of a prolific animal life.

Typical grasses are *Spartina alterniflora* and *Distichlis spicata*, which are often the dominant and only plants. In Brevard County this association can be seen in the Merritt Island Wildlife Refuge and some other limited areas near the sea.

Characteristic of this habitat are many species of aquatic birds, especially the unique clapper rail. Reptiles adapted to this salty environment are Clark's water snake, a turtle called the diamond-backed terrapin and alligators. Mammals found here include the round tailed muskrat and the marsh rabbit.

Most of the Brevard salt marshes have been diked in order to isolate them from their supply of salt water for mosquito control purposes.

FRESH WATER CANALS AND DITCHES

These important aquatic habitats are usually created for the purpose of draining the surrounding land. They are dug with draglines which deposit the soil in mounds along but one side of the ditch or canal. Common species of aquatic plants found here include water-lilies (Castalia ordorata) and water-hyacinths (Piaropus crassipes). The latter species often grow in profusion due to over-enrichment (eutrophication) of these waters. Hyacinths have few natural "enemies", since they are not a native plant species. In some places the long root-systems of the penywort (Hydrocatyle umbellata) form floating rafts of vegetation. The drainage function of the canal is often impaired by these plants and by the rapid accumulation of silt and muck. This calls for the use of the herbicide, 2-4-D, or for another dragline operation.

The small species of fish are such an obvious and interesting component of this community that a detailed description of the fauna is included here. Many of the fish species are important to mosquito control, since they feed upon mosquito larvae. Most of these fish are members of the same family of live-bearing (ovoviviparous) fishes related to guppies. All of these species are from 1/2 - 3 inches in length and show a great deal of sexual dimorphism (difference). The males of the species are usually small and slender and have the anal fin folded into a gonodopodium or mating organ. The females have a normal anal fin, but the adult females have large abdomens as a result of the presence therein of eggs and/or embryos. These species include the following:

1. Mosquito fish (Gambusia affinis): Length, 2 inches; color, gray above to white below; all populations have a few mottled mutant males; food, mosquito larvae, small crustacea; very common.

2. Sailfin Molly (Molliensia latipinna): Length, 3 inches, color, females with a yellow belly, males with a beautiful blue tail and very large dorsal fin; black mottled mutants are frequent; food, strictly algae eaters; very common.

Least killifish (Heterandria formosa): Length, 1 inch, males 1/2 inch (the tiniest of fishes); color, olive-brown with a darker stripe down each side and with traces of vertical bars; food, mostly animal; very common in dense "weedy" grass.

Other fish species lay eggs which develop in the water and are, therefore, oviparous. These include bream (bluegill), gar, eel, and smaller species as follows:

1. Flagfish (Jordanella floridae): Length, 2 inches; color, a beautiful rainbow of green, orange and yellow with a dark spot on each side; usually common.

2. Golden top minnow (Fundulus chrystus): Length, 2 1/2 inches, color very pale olive-brown with beautiful gold flecks on the sides; food, a predaceous species; usually common.

3. Red-finned killifish (Lucania goodei): Length 1 1/2 inches; color, a bold dark stripe along side from the eye to the base of the tail, fins with red blotches; usually common.

Some canals also have a great abundance of other animals including otter, leopard frogs, softshelled turtles, and mud turtles, and banded water snakes (*Natrix sipedon*).

Many of the "moccasins" seen at canals are really the non-poisonous *Natrix*. They can be told from the poison pit viper by the lack of an infrared sensitive pit located between the nostril and the eye, the presence of round, instead of slit-like pupils, and the presence of a double band of subcaudal (tail) scales. There are many harmless species of water snakes and these should be protected, since if for no other reason they eat the young of the poisonous moccasin.

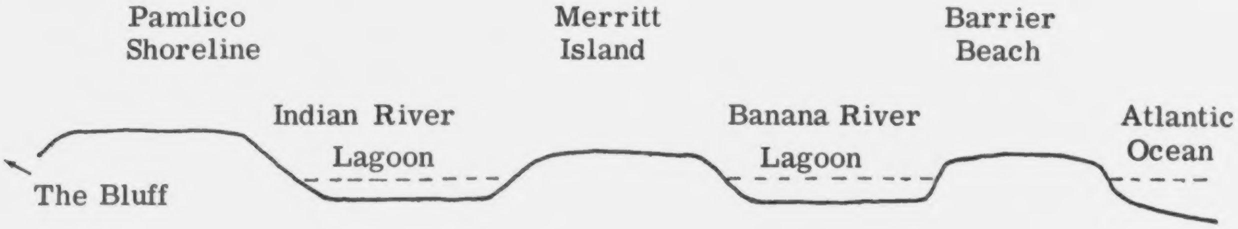
References:

Carr, Archie and Goin, Coleman, *Reptiles, Amphibians and Fresh-Water Fishes of Florida*, University of Florida Press Gainesville, 1966.

Carr, Archie, *A Contribution to the Herpetology of Florida*, University of Florida Press, 1940.

BEACH DUNES

Our ocean beach did not always reside in its present location. In the very recent geological past the ocean shoreline was further inland and in most areas in Brevard County it was located along the west bluff of the Indian River. As the sea level dropped, sand bars formed in the area of our present beaches to capture two lagoons, the Indian and Banana Rivers, and form a barrier beach.



The differences between the plant community along the west bluff of the Indian River and along the present Atlantic beach probably represent the kinds of change (succession) that occur on beach dunes.

The dunes along our present Atlantic beach have the following characteristic plant zones: the sea oats zone, the palmetto zone, and the scrub zone. The front of the foredune* is dominated by pioneer species like sea oats. railroad vine and the silver-leaf croton. In the palmetto zone, usually beginning behind the foredune, we find saw-palmetto, sea grape, wax-myrtle and red-cardinal. Further back from the sea is the scrub zone

* foredune = the first in a series of dunes, the dune closest to the sea.

which can grade into climas hammock. Here we find various scrub oaks, gopher apple, stagger bush and love vine, a parasite. Animals of the beach dunes include a lizard, the six-lined race runner, native species of mice and rats, the scrub jay, and ghost crabs. Even the female loggerhead sea turtle invades this community each June, laying her clutch of eggs above the high water mark usually on the face of the foredune.

Scientific Names:

Saw-palmetto	*Serenoa repens*
Sea oats	*Uniola paniculats*
Sea grape	*Caccalobis uvifera*
Railroad-vine	*Ipomoea Pes Caprae*
Wax myrtle	*Myrica cerifera*
Red Cardinal	*Erythrina arbarea*
Gopher apple	*Geobalanus oblongifolius*
Stagger bush	*Xalisma fruticasa*
Love vine	*Cassyth filiformis*

REFERENCE:

Kurtz, Herman, *Florida Dunes and Scrub Vegetation* (Bulletin #23, State of Florida Department of Conservation, Tallahassee, 1942)

THE OCEAN BEACH: THE TIDAL ZONE

The beach is undoubtedly the longest, narrowest, and most easily distinguished of all of the communities. It is a ribbon of sand alternately inundated and then uncovered by the oceans tides, twice each day. The action of the waves keeps the tidal zone free of terrestrial vegetation while the dessicating action of the sun and air during low tides limits the abundance of marine algae. However, despite these limitations, a surprising variety and abundance of living things inhabits the beach.

In rocky areas* the marine algae cling to the substrate with well adapted holdfasts. Sessile species found on our rocky shores include a cone-shaped mollusk, the limpet; a cnidarian, the sea anemone, and an echinoderm, the starfish.

Most of the beach is sandy, however and many animal species have adapted themselves to this plastic environment. Of particular interest is a small multi-colored oval bivale mollusk called a coquina (Donax variabilis). This species and the sand flea, (Hippa talpoida) prefer the wave washed area of the beach. Here the wave action provides these species with a constant supply of planktonic food. Both the coquina and the sand flea are a source of food for larger predators particularily shore birds and fish. The shorebird species include the ruddy turnstone, the sanderling, and the knot. These birds are often seen pursuing their specialized feeding habit, running up and down the beach following the wave action.

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*Most of the rocky area on our shores are composed of coquina, a cemented conglomerate as sand and sea shells. This is a geologic formation of the Pleistocene that extends for 150 miles along the east coast of Florida. A small bivalve mollusk of the sea shore is also known as a coquina.

Another source of nutrition on the beach is wrack. Wrack consists of a great variety of dead and dying marine plants and animals cast up by the sea; it is also contributed to by upland vegetation that has been washed into the sea. Some commonly occuring living things contributing to wrack include the massive floating brown algae, Sargassum, a great variety of dead fish; the long cigar-like seeds of the mangrove tree and the colonial cnidarian, the Portugese man-of-war. Wrack attracts scavengers such as the nocturnal ghost crab (Ocypode albicans) and a variety of insects which feed on the dead organisms. The scavenger insects attract predaceous insect species, the most prominent being the tiger beetle, a beach species with large mandibles and wings well suited for rapid controlled flight and is a common sight on warm summer days.

Man has become a dominant feature in life on the beach. He increasingly uses it for sunbathing, recreation, and even as a thorough-fare for motor vehicles. How will this effect the other living members of this complex community? We do not know.

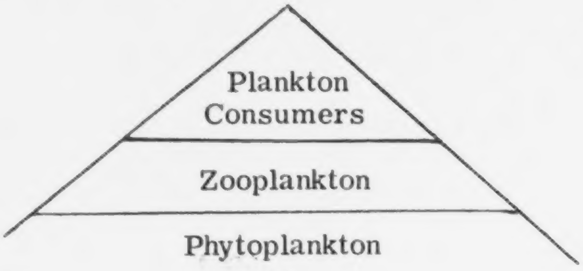
OCEANIC REGIONS

From the shore line the ocean floor slopes gently away from the land forming the continental shelf. On the Atlantic coast the width of this shelf is over 100 miles at Jacksonville, Florida; it grows progressively narrower to the south being about 26 miles wide at Cocoa and only a few miles in width at Miami. Two major habitat zones can be distinguished in this region, the neritic zone, comprising the open water above the continental shelf, and the littoral zone, the sea floor from the shore to the edge of the continental shelf. The depth of these waters varies from a few feet near the shore to over 100 feet at the edge of the continental shelf.

THE NERITIC ZONE

Most of the neritic zone allows the passage of sufficient light to support numerous phytoplankton, passively drifting or floating organisms that carry on photosynthesis. Phytoplankton includes unicellular forms, the glass walled diatoms, and the dinoflagellates with flagella that lie in grooves. (One of the dinoflagellates, Gymnodinium brevis is the organism that causes red tides.) Phytoplankton are the producers of the marine world providing the productive organic base of this ecosystem. The initial consumers of these microscopic forms are marine zooplankton which includes adult crustacea (copepods), larval invertebrates, and protozoa. Secondary consumers of plankton include larger invertebrates, small fish and even the giant baleen whales (sperm whales) which depend upon straining the plankton from the sea as their primary source of food.

Oceanic
Food Pyramid



Another planktonic form is the Portuguese man-of-war (Physalia). The top of this coelentrate is a gas filled float or sail. This is not a single organism, but a colony of small polyps many of which are armed with extremly toxic stinging cells. Physalia stands between the plankton and the nekton which comprise actively swimming animals capable of changing their positions at will.

Nekton includes fishes, whales, porpoises, sharks and rays. In our coastal waters common fishes are mullet, tarpon, Spanish mackerel, blue fish, red snapper, and grouper. The mullet is an herbiverous species feeding upon marine algae; the others are carnivorous feeding upon small fish and crustacea. All of these species are important food fish for man with the exception of the tarpon. The blue fish, for example, shadow schools of migrating baitfish--primarily menhaden and fingerling mullet. The blues group in large schools slashing their way through these fish, tearing them to bits, leaving little more than froth of tidbits and crippled fish for dipping, diving terns, gulls and pelicans that follow overhead.

THE LITTORAL ZONE

The littoral zone offers a variety of environments for diverse marine forms. The Florida littoral zone is typically sandy. These sandy shores are the result of degradation of pre-existing geologic forms and sediment transport to a site of deposition. Such sandy accumulations can take the form of reef beaches of coastal beaches of wide expanse.

Coastal beaches which typify the expanse of the Florida Coast are characteristic of areas in which the continental shelf is wide and areas in which a pre-existing coastal plain has been well established. Sedimentation and erosion processes in such areas are sensitive to seasonal winds and changes in inshore water conditions.

Marine populations in these areas are usually high in density, but due to their nocturnal behavior, are not generally observable. Small crabs, shrimp, snails, and annelid worms dwell above the surf zone; offshore, beyond the surf zone, much the same populations are represented. However, foraminifera (protists with calcareous walls) become a vital part of the benthic fauna, along with oysters, clams, snails and polychaete worms. These forms in turn provide food for higher order consumers such as shrimp fish, whelks, and starfish. An array of saprovores and scavengers, such as the sea urchin and sand dollar, feed on the remains of various marine forms.

These myriad forms of marine life seek shelter among the marine grasses which grow in regions with a stable substrate. In the deeper, cooler littoral regions, where sunlight penetration is slight, the brown alga (Phaeophycea) is the most conspicuous producer; the dark vegetation offers an ideal sanctuary for the more fulnerable life forms.

The reef beach, a second form of sand accumulation, is restricted to Florida's lower east coast. From Canova Beach to Biscayne Bay, heavy seas keep substrate stirred up. This turbid water, accompanied by a water temperature of 23-25 degrees Centigrade, offers an ideal environment for the reef-fuilding worm, Sabellariidae. This colonizing worm affixes to rocks or a firm substrate along a shoreline, forming reefs up to two feet thick. This reef-building activity may have played an important part in accumulating sand in the geologic past; thus the embryonic beginning of a chain of islands along Florida's coast. The reefs formed by the sabellariid worms provide a well protected environment for the spiny lobster, crab, sea urchin, and fish.

The sabellariid is a highly segmented worm with a long cylindrical tail. The anus, located at the distal end of the tail, can be protruded from the tube's opening to discharge wastes. The first pair of feet, which are the largest, extend forward above the head; together, these feet form a stopper for the tube entrance. Along the flattened extremities are rows of stiff spined bristles which serve as a form of protection. Each body segment has a foot on either side, enabling the worm to move up and down inside its tube. Along the back, the gills are arranged in two parallel rows. The sabellariid worm collects sand grains and shell fragments which it cements together with a secretion from a glandular area behind the mouth. Thus whole reefs are constructed through the tube-building activities of thousands of sabellariid worms.

Reproduction in the sabellariid is sexual; thousands of eggs and sperm are released. Upon fertilization, the egg develops into spherical larvae. This planktonic form is equipped with two tufts of long spiny bristles for protection. These larval forms feed on minute swimming plants. During this stage of development, cilia are the primary means of propulsion. As the larva develops, it acquires the ability to cling to rocks and construct tubes; the larvae are able to detect the presence of the glandular cement characteristic of their own species. Thus large numbers will accumulate in one area and construct sizable reefs. The long settlement period of 4-7 months is a prime factor in guaranteeing the survival of the species.

Once settled in their tubes, the sabellariid worms spread their tentacles outside the tube's opening to capture plankton for food. These tentacles are also used to seize sand grains which are then cemented together at the tube's entrance, thereby lengthening the tube and ultimately the overall size of the colony. Once this mature stage has been reached, the sabellariid worm is considered a sessile filter feeder, as it remains attached to the ocean floor for the furation of its life span and filters plankton from the surrounding water.

MANGROVE SWAMP

Mangrove swamps are found in coastal regions throughout the tropics and semitropics. In Florida, mangroves were once common along the estuaries and bays from Key West to the delta of the Suwannee River on the west coast and as far north as Brevard County and Merritt Island on the east coast. Much of its former prevalence has been limited by the development of these coastal regions.

The dominant tree is the red mangrove (Rhizophora mangle) which is easily distinguished by the presence of numerous arching aerial roots and a red-brown bark. The seeds are also unusual, since they are 10-12 inches long, pencil-shaped and germinate while still attached to the parent tree. Red mangroves in Brevard County are rare bushes; however, in the great mangrove swamps of the Everglades National Park some of the trees are 70 feet tall and up to 4 feet in diameter. In tidal regions these trees stabilize and build land by holding wave-borne debris. Red mangroves grow in areas extending both above and below the high and low tide water mark. On somewhat higher ground the black mangrove (Avicinia nitida) is found. This species sends numerous pneumatophores; aerating, erect finger-like roots above the soil surface. Other species common on wet salty soils are buttonwood (Conocarpus erectal), cocoa-plum (Chrysobalanus icaco), saltwork (Batis maritima), sea grape (Coccolobus uvifera) and saltbrush (Baccharis spp.).

Characteristic animals of this community are the salt water banded water snake, Clark's water snake, the coconut crab, and the diamond-back terrapin, a turtle. In the Everglades National Park the alligator and crocodile are both members of this ecosystem. Raccoons, rat snakes, glass "snakes", fiddler crabs, and numerous species of fish are prevalent. In Brevard County this association is found along the east shores of the Banana and Indian Rivers, especially along the entrance road to Long Point County Park.

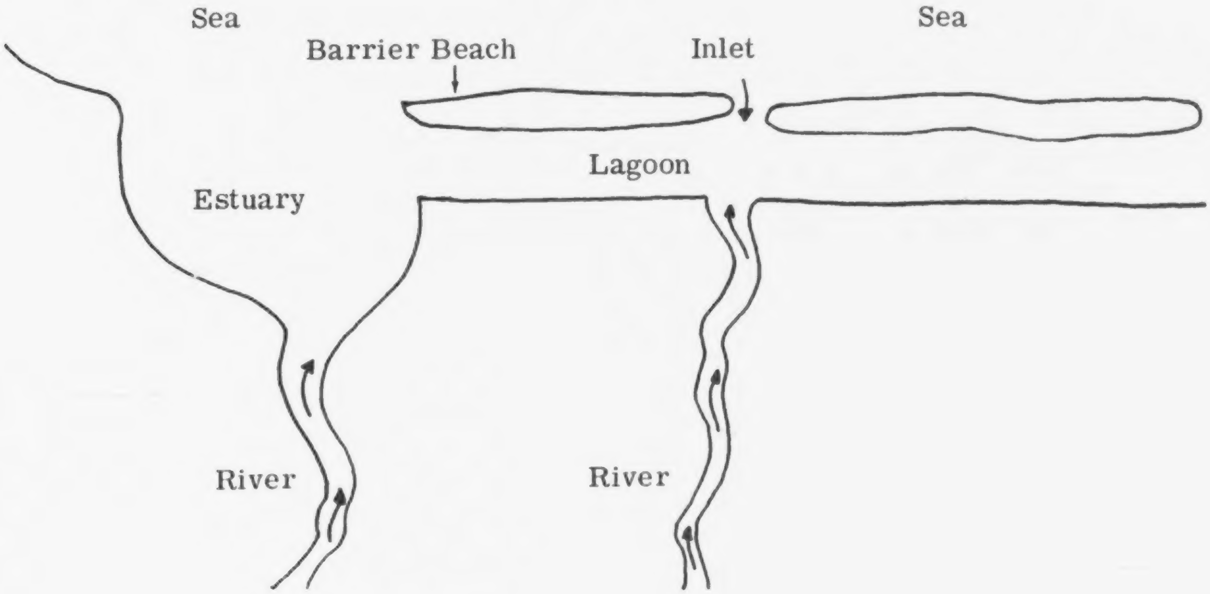
ESTUARIES AND LAGOONS

Estuaries and lagoons have relatively shallow brackish water resulting from the mixing of fresh water and salt water in regions where rivers meet the sea. An estuary is defined as the wide mouth of a river where the tide meets the river's current, while a lagoon is salty water parallel to, and separated from, the sea by a barrier beach. During high tides water enters the estuary or lagoon from the sea, often rushing under the fresher water which floats on top of the incoming sea water. The outgoing tide reverses this process carrring brackish water back out to sea.

The waters of estuaries and lagoons are highly productive of marine life which abounds in these waters. One reason for this abundance is the mixing of nutrients, carried from the land by rivers and streams, with sea water resulting in the growth-promoting environment known as brackish water. These waters are the sheltered nurseries for a host of marine fish of considerable value in sport and commercial fisheries. The basis of this productivity is marine algae both planktonic (free floating) and sessile or attached types. Filter-feeding shellfish, oysters and clams in particular, grow rapidly on the microscopic algal plankton. Algae-eating fish, such as mullet, subsist upon sea lettuce and other sessile species. Even the sea cow, a marine mammal, is completely dependent upon marine vegetation as a source of food.

The predators of the estuaries and lagoons include sea trout, pelicans, ospreys, sting rays and sharks. These animals subsist upon small fish and a variety of crustacea including shrimp. The estuaries provide quiet shallow waters for the breeding and young of shrimp, sea trout, mullet, snook and even sea horses.

The waters also produce an abundance of food for millions of migratory waterfowl. These include, scaup, grebe, mergansers, coots and cormorants. Most waterfowl are migratory and appear in the lagoons and estuaries during the fall, winter, and spring--summers are spent breeding in the wet prairies and tundra of Canada.



* In Brevard County our lagoons are the Banana and Indian Rivers.

INLETS

Inlets, which are channels (often man made) connecting the Indian River with the ocean, offer an avenue of exchange; high tides sweep fresh ocean waters into the river and enables an exchange of nutrients and wastes. Such tidal waters may also bring various life forms which require the estuarine waters to complete their reproductive cycle. At low tide various larval forms may be returned to the ocean to complete their maturation or serve as food for higher order consumers.

The inlet's importance as an avenue of interaction between the sea and the river estuary is a dimension sometimes overlooked as the inlet is usually perceived as a convenience for man's commercial and recreational activities. Inlets must be deep and wide enough to allow freighters as well as outboards passage into a given port. Periodically this waterway must be dredged to ensure its continued usefulness to man. The inlet's depth may decrease due to an accumulation of sediments which may be introduced into the water by the erosive action of rain and waves. Another contributor is the remains of various marine forms of flora and fauna. The percentage that the latter contributes will be influenced by the "health" of the estaurine waters. If large amounts of sewage are introduced, the accumulation will increase.

To deter the accumulation of sediment, large rocks similar to those lining the edge of the inlet, may be layered along the bottom of the inlet. Not only do these rocks deter sedimentation, but also offer a suitable substrate for such marine life as sea urchins and sponges.

Though rocklined inlets may be viewed as purely functional routes to the sea, the ecosystems sheltered here are much more complex than a superficial glance might suggest. The rocks defining the margin of the inlet, as well as various marker buoys and channel markers offer a firm substrate for various marine organisms to attach. Plants represented are chiefly green algae, brown algae, and seaweeds adapted to the hostile environment created by a ceaseless rise and fall of tide waters. This firm substrate also offers an ideal environment for a number of marine animals, the majority of which are sessile filter feeders. These creatures remain attached to the rocky surfaces for the duration of their mature life, feeding on various plankton forms which swim freely in the surrounding waters. These sessile marine forms face possible dessication if low tides expose them to the drying influences of the sun and wind.

An example of a sessile filter feeder which has adapted to the hostile environment created by changing tides is the barnacle, Balanus balanoides. This familiar marine form is often found resting on wharf pilings and hulls of boats. It is shielded by an external calcareous shell which seals in moisture when the tide ebbs. When the tide again rises, this shrimp-like creature opens its shell-armor and extends its legs into the surrounding water to capture its prey.

Another well protected neighbor of the barnacle is the limpet; it, too, has an external shell, conical in shape, for protection in a hostile environment. Unlike the barnacle, the limpet's flattened conical shell has no opening; in fact, it fits so firmly to the substrate that an operculum is unnecessary. Moisture is retained in a groove circling the interior of the shell, thus, the gills are kept moist until the tide return. Another distinguishing feature of the limpet is its diet which consists of marine algae growing on the wave battered rocks. This primitive mollusk is equipped with a long radula or tongue which enables it to scrape off algae as it moves slowly over the surface of the rocks.

A third form of marine fauna that may inhabit this environment bears a stronger resemblance to a plant, rather than an animal; this is the sea anemone. It's deceptive flower-like appearance is produced by a cluster of tentacles at the distal end of a long slender cylindrical solumn. These tentacles are simple, hollow, and taper to a point or a ball-like enlargement. The tentacles release microscopic "darts" which secrete a substance which has a paralyzing effect upon microscopic prey. The stunned prey are swept into the mouth, a hollow slit-like pore in the center of a clear, smooth zone, separating it from the tentacles. The base, located at the proximal end of the cylindrical tube, enables the sea anemone to stay firmly anchored to the hard substrate; this base is quite versatile in that it also enables the sea anemone to move. The sea anemone has no protective external shell or supportive skeleton. The larvae are free swimming, as are the larval forms of the previously mentioned marine animals. It may reproduce sexually or asexually by means of fission or budding. Usually those creatures are found on rocks protected from direct wave action.

After a cioser look, it is obvious that the inlet offers a scenario of of marine life forms to investigate. For the curious ecologist, it is an easily accessible way of observing marine life, undistrubed in marine environment; an opportunity surpassed only by tidal pools characteristic of northern, rocky coastlines.

CYPRESS DOME COMMUNITIES

GENERAL DESCRIPTION AND LOCATION:

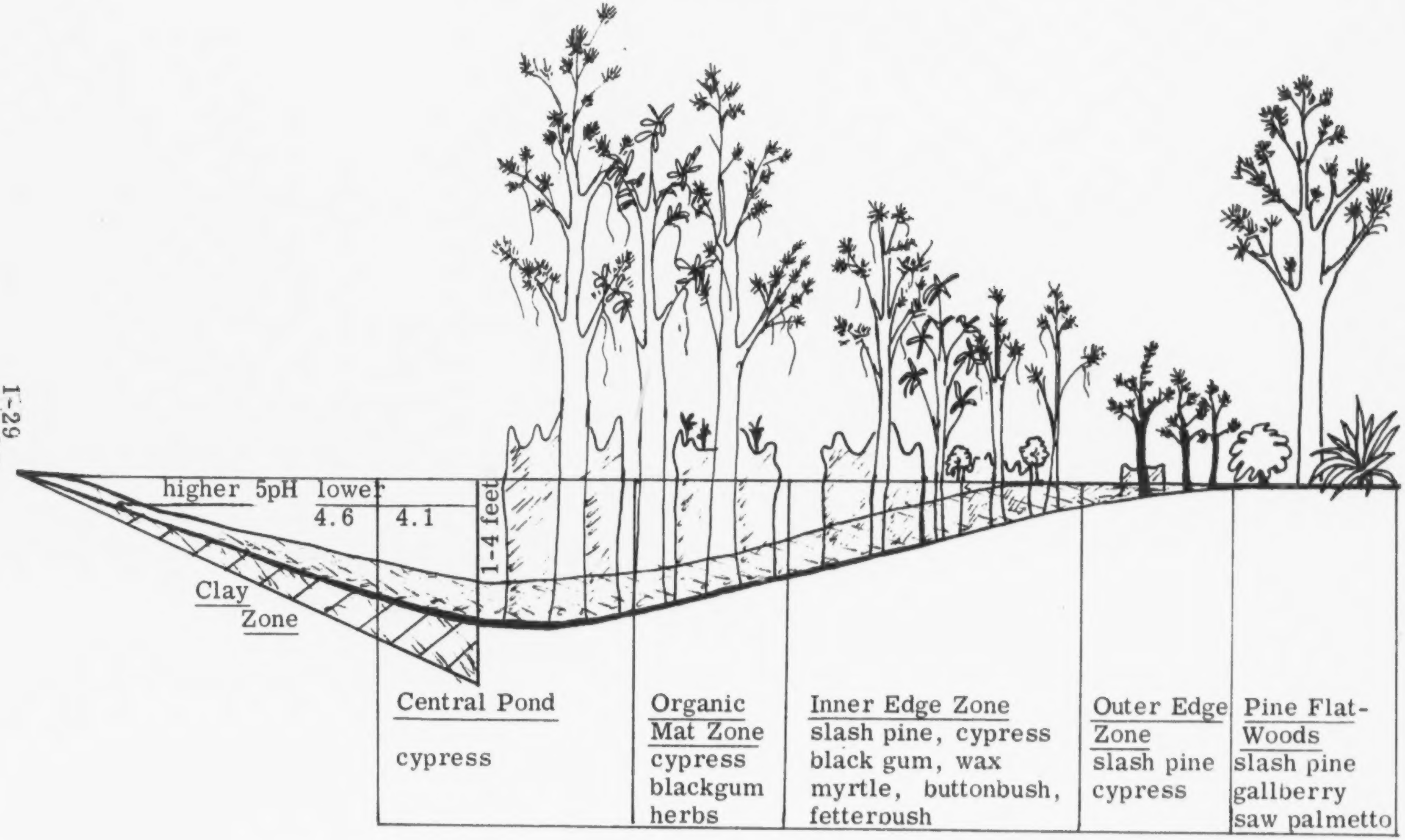
Among the pine flatwoods of the Gulf and southern Atlantic coastal plains, a distinct stand of vegetation can be observed breaking the horizon. These stands are actually ponds of water conspicuously populated with pond cypress (*Taxodium ascendens*). More often than not, they are characterized by a symmetrical dome-like shpe which is the result of larger trees growing in the center and smaller ones on the periphery. These distinct communities are commonly referred to as "cypress domes" or "heads" and have always been an area of interest, particularly since cypress building materials have long been known for durability against decomposition.

Cypress domes, along with bayheads and gum swamps, occupy the wettest, poorly drained depressions of central peninsular Florida. Although fire subclimzx pine flatwoods makes up about 50% of the total land area, the areas of swamp forest make up a distinct ecological situation. Some ecologists suggest that these swamp environments form a successional between the flatwoods and the climas hardwood hammocks.

FLORAL AND PHYSICAL DESCRIPTION

Cypress heads have a unique internal structure. The center is composed of a pond of stagnant acid water that varies from 1 to 4 feet deep. It is here that the cypress trees are the oldest and tallest, although they may not occupy the exact center. Very little debris collects on the

CROSS SECTION OF A CYPRESS DOME


higher 5pH lower
4.6
4.1
1-4 feet
Clay Zone
Central Pond
cypress
Organic Mat Zone
cypress
blackgum
herbs
Inner Edge Zone
slash pine, cypress
black gum, wax
myrtle, buttonbush,
fetteroush
Outer Edge Zone
slash pine
cypress
Pine Flat-Woods
slash pine
gallberry
saw palmetto

smooth buttress roots, consequently, very little other vegetation occurs here. This central pond, in well established heads, rarely dries up even in the most severe dry seasons.

The second zone, called the organic mat zone, occurs at the edge of the central pond. Here the water is shallower, becoming more so toward the edge of the dome. This zone is characterized also by large cypress, although some smaller ones may be found along with a few large black gums. The cypress buttresses here have considerable accumulation of organic debris, which forms mats, as a result many small woody and herbaceous plants grow here.

The largest zone of the head lies between the organic mat zone and the outer edge. This area is characterized by considerable fluctuation in water depth. During the rainy season, the depth may reach 2 feet, but may drop to zero during the during the early spring. Here also, cypress of all sizes may be found along with many black gums and slash pines. Other plants occurring here are white bays, swamp red bays, and red maples. A distinct understory of wax myrtle, button bush, and fetter bush can usually be found. Beneath the understory, clumps of Virginia chain fern, lizard's tail, red-root and other herbaceous plants occur.

The last zone is referred to as the inner edge. Here almost all the cypresses are of sapling or seedling size. Small slash pines and hardwood saplings may also be found. These saplings contribute to the dome-like appearance of the head. The ground cover consists of chain ferns, wire grass, and saw grass, but many flatwood species may also be found. It is only during the wet summer months that the surface may be covered with water.

Around the outer edges of the cypress heads, one finds the typical flatwoods vegetation, pines, gallberry, and saw palmetto.

Several important environmental factors should be mentioned. One is pH. As mentioned earlier, hydrogen ion concentration increases toward the center of the dome. So do the more mature cypress trees, which reflects a strong relationship between low pH tolerance and that species. This tolerance does not appear to be true of other species.

The second factor is the character of the soil. All cypress heads are underlain with marine deposited clay. This clay is deeper toward the center, consequently the water is deeper there. Cypress knees or buttresses are thought to be for structural adaptations gas exchange, therefore the cypress again appears to be more tolerant of the water than the other species. The presence of this clay deposit in depression occuring in the flatwoods appears to be the precurser of the cypress dome. It is thought also that the precurser plant is sphagnum moss, which may be found in mats near the center. Sphagnum moss contributes to the acidity, thus probably paving the way toward a favorable environment for cypress survival.

The third factor is fire which is a distinct environmental factor of the surrounding flat woods. Since the water level is low there, the outer edges become susceptible to the devastating winter fires and consequently one finds the cypress there stunted or destroyed by fire. Along with the prior mentioned factors, low pH, standing water because of the deeper clay, fire seems to contribute to the distinct symmetrical shape of the cypress dome.

Other factors contributing to the floral structure of the domes are the minerals calcium, phosphorous, and magnesium, and the organic matter content of the soil. Below is a cross section of a typical dome, showing zonation and some of the important influencing factors. (See diagram of cypress dome.)

ANIMALS

Very little seems to be written concerning the animals found in cypress domes. However, since the pine flatwoods become dry during the winter season, one can assume that the animals typically found in the surrounding flatwoods use the ponds as a source of water. One animal does seem to prefer this habitat; the cottonmouth water moccasin. Other frequent visitors are the red-shouldered hawk, snapping turtles, wood ducks, raccons, opossums andothers. Here lies an opportunity for study for students in Ecology.

THE FUTURE OF CYPRESS DOMES

Since cypress is still harvested, there are probably few stands that do not show the influencing factor of the presence of man. Also recent ditching,draining, and clearing operations for the production of more profitable slash pine, and for pasturing cattle, have resulted in the total destruction of many stands. Others are beginning to show the signs of man's influence at an alarming rate. Should these distinct ecological units be totally destroyed or do they play a not yet understood role in the intricate balance of nature in Florida? This question is an important one and there may not be time enough to answer it before the consequences are known.

REFERENCE:

Monk, Carl D. and Timothy Brown, "Ecological Consideration of Cypress Heads in North Central Florida," American Midland Naturalist, Vol. 74, No. 1, July, 1965.

THE HIGH-PINE COMMUNITY

The high-pine country contains an association of living things adapted to a rolling well-drained topography. This is an open pine forest with sandy soils well suited to the burrowing habits of a variety of animals and the growth of plants requiring deep porus soils. The dominant tree, the long-leaf pine (Pinus australis), has been greatly reduced in number due to its value as a timber tree, its susceptability to fire, and clearing of the high-pine for agriculture purposes. Most of central Florida's orange groves have replaced these piney hills with a characteristic assemblage of grove organisms. Scattered beneath the long-leaf pine are a variety of small oaks including the common turkey oak (Quercus laevis), the blue-jack oak (Q. cinerea), and twin oak (Q. geminata). The lower-level plants include saw palmetto (Serenoa repens), wire grass (Aristida stricta), and gopher apple (Geoblanus oblongifolus).

One of the most characteristic elements of the fauna is the fence-lizard (Scaphopus holbrookii), a small grey or tan lizard with blue patches on its throat. Burrowing species include the gopher tortoise (Gopherus polyphemus) and a small rodent, the pocket-gopher. Both of these fossorial* species leave their characteristic marks on the landscape. The tortoise burrows are easily recognized since they have large open mouths fronted with mounds of sand. The pocket-gophers build smaller mounds of sand which completely block entrance to the burrow. Both of these

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*Fossorial - adapted for digging or burrowing.

"gophers" are important herbivores; the tortoise eating portions of plants above the soil while the rodents select the roots of plants beneath the soil surface. Predator amphibian species include the pine-woods tree frog (*Hyla femoralis*), the bell tree frog (*Hyla gratiosa*), andthe gopher frog (*Rana capito*), a large mouthed species that lives in tortoise burrows, and the tiny toad (*Bufo quercicus*), who makes small burrows of his own. Some larger animals are the coach-whip snake (*Coluber flagellum*), the pine snake (*Pituophis nugitus*), skunks, red-bellied woodpeckers, barred owls, and foxes.

This community occurs most commonly in central and north Florida, but is found in only scattered locations in south Florida and along the coast.

MERRITT ISLAND NATIONAL WILDLIFE REFUGE

Merritt Island National Wildlife Refuge, Brevard County, Florida, was created on August 28, 1963. It contains 140,393 acres. It is administered by the Bureau of Sport Fisheries and Wildlife in the U. S. Department of the Interior. It was established as a sanctuary for wintering and migratory waterfowl through an agreement between the Department of the Interior and the National Aeronautics and Space Administration. Merritt Island Refuge serves as a buffer zone between the mainland and the sites at the Kennedy Space Center.

The types of wildlife habitat vary greatly. Specifically, the refuge is composed of the following: brackish marsh - 48%, palmetto - upland pine - 26%, ocean beach - 5%, oak hammocks - 5%, citrus groves - 5%, former housesites with exotic vegetation - 5%, mangrove islands - 4%, and fresh water marsh - 2%.

Several endangered wildlife species are present at the refuge among the hundreds of other species which are not in danger of extinction at this time. Some of the endangered species are the southern bald eagle, American peregrine falcon, dusky seaside sparrow, Florida manatee or sea cow, and American alligator. The refuge staff manages the land and the wildlife. At the present time, two biological studies are in progress; one deals with southern bald eagle reproduction and the other concerns the natural history and especially the habitat requirements of the dusky seaside sparrow.

The refuge is the site of several types of recreation including general nature study; birdwatching; conservation-oriented youth group camping; salt water, surf, and fresh water fishing; waterfowl hunting; and swimming.

The Merritt Island Refuge is planning for its future needs now. One of its top priorities is the creation of study facilities for environmental education classes. It is hoped that within the next few years it will be able to provide study sites located in various plant communities to allow students to better understand their natural environment. At the present time, to meet environmental education needs, refuge staff members give nature walks through different parts of the refuge. Refuge staff members also talk to classroom groups upon request.

More information regarding the refuge is available from the Refuge Manager, Merritt Island National Wildlife Refuge, P. O. Box 6504, Titusville, Florida 32780.

BIRDS OF THE MERRITT ISLAND NATIONAL WILDLIFE REFUGE

Merritt Island National Wildlife Refuge was established in August 1963 by agreement between the Bureau of Sport Fisheries and Wildlife and the National Aeronautics and Space Administration. The refuge occupies the buffer zone adjacent to missile launching sites on Cape Kennedy and the John F. Kennedy Space Center.

Its 46, 530 acres are composed of shallow, fresh water impoundments and saltwater creeks and lagoons. Scattered stands of slash pine, palmetto, oak, and cabbage palm are found on the higher elevations.

The mottled duck is resident on the area and nests in spring and summer. Thousands of ducks comprising more than 20 species winter on the refuge, some arriving by late August and remaining until April. Lesser scaup are present in the largest numbers, followed by widgeons, pintails, and blue-winged teal Coots are present in numbers far greater than those of all ducks combines. An occasional snow or blue goose is observed.

Numerous wading birds and shore birds are present throughout the year, many nesting on or near the refuge. During the winter the brown pelican and the larger white pelican are commonly observed. The southern bald eagle nests on the refuge and generally can be observed without undue difficulty from November through March. The refuge provides some of the little remaining habitat for the rare dusky seaside sparrow. Both it and the eagle are found on the official endangered species list.

The following list of 224 species represents observations made by refuge personnel, members of the Indian River Audubon Society and other visitors since 1951. An additional 31 species or casual or accidental occurrence are listed in an appendix. Those species preceded by an * are known to nest on the refuge. The list is in accordance with the Fifth (1957) A. O. U. Check-list. Symbols used are:

S - March-May
S - June-August
F - September-November
W - December-February

a - abundant
c - common
u - uncommon
o - occasional
r - rare

	S	S	F	W
Common Loon			u	u
Horned Grebe			c	c
*Pied-billed Grebe	a	a	a	a
White Pelican	c	u	u	c
Brown Pelican	c	c	c	c
Gannet				r
Double-crested Cormorant	c	c	c	c
*Anhinga	c	c	c	c
Magnificent Frigate-bird	o	o	o	o
Great White Heron		r		
Great Blue Heron	c	c	c	c
*Green Heron	c	c	c	c
Little Blue Heron	c	c	c	c
Cattle Egret	c	c	c	c
Reddish Egret		r		
Common Egret	c	c	c	c
Snowy Egret	c	c	c	c
Louisiana Heron	c	c	c	c
Black-crowned Night Heron	c	c	c	c
Yellow-crowned Night Heron	u	u	u	u
*Least Bittern	c	c	c	c
American Bittern			u	u
Wood Ibis	u	u	u	u
Glossy Ibis	u	u	u	u
White Ibis	c	c	c	c
Roseate Spoonbill		r	r	
American Flamingo		r		
Canada Goose				r

	S	S	F	W
Snow Goose				r
Blue Goose				r
Fulvous Tree Duck				r
Mallard	o	o	u	u
Black Duck			u	u
*Mottled Duck	c	c	c	c
Gadwall			c	c
Pintail	u		c	c
Green-winged Teal	u	r	c	c
Blue-winged Teal	c	o	c	c
American Widgeon	c	o	c	c
Shoveler	c	o	c	c
Wood Duck			u	u
Redhead			u	u
Ring-necked Duck			c	c
Canvasback			u	u
Greater Scaup			u	u
Lesser Scaup	c	o	a	a
Bufflehead			u	u
Oldsquaw			r	r
Ruddy Duck	u		c	c
Hooded Merganser			u	u
Common Merganser				r
Red-breasted Merganser	u		c	c
*Turkey Vulture	c	c	c	c
Black Vulture	o	o	o	o
Swallow-tailed Kite	r	r		
Sharp-shinned Hawk			o	o
Cooper's Hawk			o	o
*Red-tailed Hawk	u	o	u	u
*Red-shouldered Hawk	u	u	u	u
Broad-winged Hawk			r	r
*Bald Eagle	u		u	u
Marsh Hawk	u		c	c
Osprey	o	o	c	o
Peregrine Falcon			o	o
Pigeon Hawk			u	o
*Sparrow Hawk	u	u	c	c
*Bobwhite	c	c	c	c
*King Rail	u	u	u	u
*Clapper Rail	u	u	u	u
Virginia Rail			r	r
Sora	c		c	c
*Black Rail	o	o	o	o
*Common Gallinule	c	c	c	c

	S	S	F	W
*American Coot	c	c	a	a
Semipalmated Plover	u		u	u
Piping Plover			r	r
Wilson's Plover	o	o	o	o
*Killdeer	c	c	c	c
Black-bellied Plover	u		u	u
Ruddy Turnstone	u		c	c
American Woodcock			o	o
Common Snipe			u	u
Whimbrel				o
Spotted Sandpiper			u	r
Solitary Sandpiper			o	o
Willet	o	o	o	o
Greater Yellowlegs	u		c	u
Lesser Yellowlegs	c		c	u
Knot				r
Purple Sandpiper			o	o
Pectoral Sandpiper			r	r
White-rumped Sandpiper				r
Least Sandpiper	u		u	u
Dunlin	c		c	c
Short-billed Dowitcher			c	c
Long-billed Dowitcher			u	u
Stilt Sandpiper			r	r
Semipalmated Sandpiper	u		u	u
Western Sandpiper			u	u
Marbled Godwit			r	r
Sanderling			u	u
American Avocet			r	r
*Black-necked Stilt	c	c		
Great Black-backed Gull				r
Herring Gull	c	c	c	c
Ring-billed Gull	a	a	a	a
Laughing Gull	c	c	c	c
Bonaparte's Gull	u	u	c	c
Gull-billed Tern	u	c		
Forster's Tern	c		c	c
Common Tern			r	r
Least Tern	u	c		
Royal Tern	a	a	a	a
Sandwich Tern			r	o
Caspian Tern			c	a
Black Tern			c	
Black Skimmer	c	c	c	c
*Mourning Dove	c	c	c	c

	S	S	F	W
*Ground Dove	c	c	c	c
*Yellow-billed Cuckoo	u	u	u	
Barn Owl	o	o	o	o
*Screech Owl	o	o	o	o
*Great Horned Owl	u	u	u	u
*Barred Owl	o	o	o	o
Short-eared Owl			o	o
*Chuck-will's-widow	u	u	o	o
Whip-poor-will	r		o	o
*Common Nighthawk	u	u		
*Chimney Swift	u	o		
Ruby-throated Hummingbird	u	r	u	o
Belted Kingfisher	u	u	c	c
*Yellow-shafted Flicker	u	u	u	u
*Pileated Woodpecker	o	o	o	o
*Red-bellied Woodpecker	u	u	u	u
Red-headed Woodpecker	r	r	r	r
Yellow-bellied Sapsucker			u	o
*Hairy Woodpecker	r	r	r	r
*Downy Woodpecker	o	r	o	o
*Red-cockaded Woodpecker	o	o	o	o
*Eastern Kingbird	u	c	c	
*Gray Kingbird	u	u		
Western Kingbird			o	o
*Great Crested Flycatcher	u	u	o	o
Eastern Phoebe	u		c	c
Tree Swallow	o		a	a
*Barn Swallow	c		c	
*Cliff Swallow	o		o	
Purple Martin	o	o		
*Blue Jay	u	u	u	u
*Scrub Jay	c	c	c	c
*Fish Crow	c	c	c	c
*Brown-headed Nuthatch	o	o	o	o
House Wren			u	u
*Carolina Wren	c	c	c	c
Long-billed Marsh Wren			o	o
Short-billed Marsh Wren			u	u
*Mockingbird	c	c	c	c
Catbird	c		c	c
*Brown Thrasher	u	u	u	u
Robin	c		a	a
Hermit Thrush			r	r
Swainson's Thrush	u		u	o
Veery	u		u	

	S	S	F	W
Eastern Bluebird	u	u	u	u
Blue-gray Gnatcatcher	u		u	u
Ruby-crowned Kinglet			u	u
Water Pipit			r	r
Cedar Waxwing			r	r
*Loggerhead Shrike	u	u	u	u
*Starling	c	c	c	c
*White-eyed Vireo	u	u	u	u
Solitary Vireo			r	r
*Black-whiskered Vireo	o	o		
Black-and-white Warbler	u		ur	
Swainson's Warbler		u		
Orange-crowned Warbler			o	o
Yellow Warbler	o		o	
*Cape May Warbler	c		c	
Black-throated Blue Warbler	c		c	o
Myrtle Warbler			c	c
Black-throated Green Warbler			r	r
Yellow-throated Warbler			o	o
Blackpoll Warbler	c		c	
*Pine Warbler	u	u	o	u
*Prairie Warbler	u	u	u	u
Palm Warbler	o		c	c
Ovenbird	u		u	o
Northern Waterthrush	u		u	o
*Yellowthroat	c	c	c	c
Yellow-breasted Chat			r	r
American Redstart	c		c	o
House Sparrow	c	c	c	c
Bobolink	u		u	
*Eastern Meadowlark	u	u	u	u
*Ring-winged Blackbird	a	a	a	a
Baltimore Oriole			r	r
Rusty Blackbird			o	o
*Boat-tailed Grackle	c	c	c	c
Common Grackle	c	c	c	c
Brown-headed Cowbird			o	o
*Summer Tanager	c	c		
*Cardinal	u	u	u	u
Indigo Bunting	o		o	o
Painted Bunting	o		o	o
Dickcissel			r	r
American Goldinch			u	u
*Rufous-sided Towhee	c	c	c	c
Savannah Sparrow	c	c	c	c

	S	S	F	W
Grasshopper Sparrow			o	o
Henslow's Sparrow			r	r
Sharp-tailed Sparrow			r	r
Seaside Sparrow			r	r
*Dusky Seaside Sparrow	r	r	r	r
Vesper Sparrow			r	r
Lark Sparrow			r	r
Bachman's Sparrow	r	r	r	r
Chipping Sparrow			u	u
Field Sparrow			o	o
White-crowned Sparrow			o	o
White-throated Sparrow			u	u
Fox Sparrow			r	r
Lincoln's Sparrow			r	r
Swamp Sparrow	o		u	u
Song Sparrow	o		u	u

The following 31 species have been recorded, but are considered of casual or accidental occurance:

Red-throated Loon
European Widgeon
Common Goldeneye
White-winged Scoter
Yellow Rail
American Oystercatcher
American Golden Plover
Roseate Tern
Sooty Tern
Noddy Tern
Black-billed Cuckoo
Smooth-billed Ani
Scissor-tailed Flycatcher
Acadian Flycatcher
Bank Swallow
Brown Creeper
Wood Thrush
Gray-cheeked Thrush
Golden-crowned Kinglet
Prothonotary Warbler
Worm-eating Warbler
Tennessee Warbler
Nashville Warbler
Blackburnian Warbler
Hooded Warbler
Orchard Oriole
Scarlet Tanager
Rose-breasted Grosbeak
Blue Grosbeak
Purple Finch
Pine Siskin

Refuge Leaflet 225
May 1967

BIOMES & ECOSYSTEMS

THE ST. JOHN'S RIVER AND MARSH ECOSYSTEM

The longest river in Florida, the St. John's River, has its headwaters here in Brevard County. Its waters originate in the marshes (Jane Green) surrounding Lake Hell'n Blazes, the first in a chain of fluvial lakes. From Lake Hell'n Blazes the river flows northward to Saw Grass Lake and then to Lake Washington, the sites of two water treatment plants that supply most of the people in South Brevard County with their supply of fresh water.

The dark coffee-colored waters, of our rivers and streams are characteristic of waters that originate on the southeastern coastal plain of the United States. The color, is due to organic acids that originate in the marginal floodplain swamps or bayheads.

The edges of the upper St. John's River are bordered with a variety of flowering plants including a beautiful amaryllis, St. John's lily, saw grass, a hibiscus species called marsh mallow (*Crineim*), shrubs such as buttonbush (*Cephatanthus occidentalis*) and willow (*Salix longipes*). The fish in the river include black bass, bowfin, gar and a variety of bream species. Reptilian species found in the river include the rare American alligator, the common Florida cooter (*Pseudemys floridana*) and a few Nelson's turtles (*Pseudemys nelsoni*). These turtles depend upon a good supply of submergent aquatic plants for food, while the alligator depends upon these turtles for a substantial portion of its diet.

Birds are usually abundant along the river. The white heron-like birds are the large American egret, the smaller snowy egret, and the inmature little blue heron. The heron with the brown bib is the Louisiana heron. Common large black birds are boat-tailed grackles and a smaller species is the red-winged blackbird. Swimming on the water surface are pie-billed grebes and coots. Mammals along the rivers' edge are the marsh-rabbit, the roundtailed muskrat, otter and racoons.

Common harmless snakes are banded water snakes (Natrix sipedon), green water snakes (*Natrix cyclopion*) and the brown water snake (*Natrix taxispilota*). The poisonous moccasin (*Ankistrodon piscivorus*) is rare along the upper St. John's River. These snakes feed upon a variety of frogs, especially the Southern bull-frog (*Rana grylio*) the leopard frog (*Rana pipiens*) and the large green tree frog (*Hyla cinerea*).

The ecology of the St. John's River basin has been severly altered. Dikes and ditches have been built around large areas of the marshland for the purpose of drainage. During periods of abundant rainfall the excess water is pumped over these dikes limiting the usefullness of these areas as natural reservoirs. Plans are now being made to construct a series of reservoirs to impound water to be released during periods of drought. This action is required since the limits of the natural marshlands are being considerably reduced for urban development and cattle. This course of action should be carefully considered before initiated for the following reasons:

(1) the relatively small reservoirs planned will eventually have to be maintained at high water levels,

(2) impounded water tends to accumulate nutrients and eutrophicate,

(3) biologists expect stratification and subsequent water degradation to result, and

(4) as long as deep reservoirs are available no restraints will be set on the further development of the marshes.

The natural marshes act as a wonderful "sink", absorbing rainfall and nutrients into the lift systems and slowing releasing the purified water to the St. John's River and lakes. The seasonal fluctuation of shallow marsh water further allows the marsh to cleanse away (oxidize) the accumulated organic load through periodic direct exposure to the air and sunlight.

FUTURE OF FRESH WATER FISHES IN FLORIDA

By

John W. Woods

Florida has a nationally famous reputation for its high quality fresh water fishing. Last year more than 152,000 non-resident fishing licenses were sold in addition to the 460,000 resident state and county licenses. It is estimated that the fishermen contributed more than $150,000,000 to the total economy of Florida in 1967-68 by purchasing items relating to the pursuit of this sport. Fresh water fishing is a resource that has played an important role in making Florida the great state that it is today.

While sport fishing is not considered in the same category as production of food and fiber, it most certainly plays a major role in the making for a desirable environment with the increasing pressure of modern life, the chance to "get away from it all" simply for recreation is taking on added importance. The future demand for fresh water fishing and related recreation will greatly increase with the expected growth of the state.

The most important problem now facing Floridians in the battle for a quality environment and desirable fresh water sport fishery is pollution. Pollution is anything that alters our environment, making it less desirable to the well being of man. When man's environment deteriorates, so does many of the creatures man considers as desirable (fresh water fish). Numerous examples of pollution and deterioration already stand out in Florida. Fortunately, we have such a tremendous wealth of fresh water lakes and streams that people were able to move to other lakes and streams when their favorite area became polluted and no longer desirable for fishing or other types of water recreation. The pace of pollution has increased at such an astounding rate in the past few years that the outlook for the future is quite dim as we are rapidly running out of new bodies of water to turn to when others are polluted.

Through geological aging, greatly accelerated by man's activity, many of Florida's lakes and streams are rapidly becoming highly eutrophic or over-enriched. Unless there is a change in the values of Florida's fresh water lakes, this trend will increase. The eventual result will be septic conditions in most of the lakes and streams in Florida. The changes in the aquatic environment from pollution are irreversible without tremendous expenditures of money and manpower. Technology available at the present time can stop and reverse some aspect of eutrophication; however, most people are not ready to accept the hardships or costs involved in such an undertaking.

Some of the major causes of degradation of aquatic habitat and fresh water fishing in Florida are as follows:

1. Discharge of domestic agricultural and industrial wastes into rivers and lakes without proper treatment. The gross examples of improperly treated waste being dumped into bodies of water destroying aquatic habitat have been presented numerous times. They would include the Fenholloway River, St. Johns River from Palatka to Jacksonville, Peace River and Lake Apopka. Less

publicized examples are even more numerous.

2. Stabilization and reduction of water levels in rivers and lakes which have historically fluctuated widely. Such fluctuation helps oxidize organic buildup and flush out some of the soluble nutrients present in the lakes. Stabilization enhances waterfront development but transforms the lake into a stagnant watershed nutrient trap which creates ideal conditions for rapid eutrophication. This not only destroys the aquatic environment for the desirable fresh water fishes, but renders the lake unsuitable for contact water sports and eventually reduces waterfront property values.

3. The increased run-off of pesticides and nutrients from the watershed. The natural marshes and flood plains which formerly acted as a "biological waste treatment complex" absorbing and utilizing excess watershed nutrients are rapidly being drained and channelized. Channelization provides an excellent conveyor of nutrients and pesticides to our fresh water lakes, streams and estuaries.

4. Unrestricted dredging and filling of lakes and streams for industrial and urban development of waterfront sites. In the last two years over 200 illegal dredge and fill operations have been reported to the Trustees of Internal Improvement Fund. Each of the operations destroyed vital littoral zone for nursery grounds required for desirable fresh water species.

5. Elimination of the full utilization of a natural resource by restricting the removal of commercially harvestable fishes. In most cases this would be beneficial and not detrimental to all interests concerned.

6. Uncontrolled killing of water hyacinths and other so called noxious aquatic weeds. By allowing the dead plants to sink to the bottom, there is a rapid re-circulation of nutrients into the water column, creation of anerobic conditions and a significant buildup of bottom silt. Each time these plants are sprayed, it increases the opportunity for the nutrients to be utilized in a less desirable form such as algae blooms.

7. Unauthorized importation and introduction of exotic fresh water fish species. Many of the introduced species compete and can replace desirable native fresh water fishes. Introduction of exotic fishes for biological weed control without consideration of ecological damage to the native fresh water aquatic habitat.

The problems of lakes and streams of Florida have been briefly described with expected effects on aquatic life. They will continue to deteriorate the aquatic environment unless there is full implementation of available and future technology. In a recent report from the Game and Fresh Water Fish Commission in which predictions were made about a sport fishing in the St. Johns Valley in the year 2020, we indicated there would be only two places in the 18 county area where largemouth bass could be caught if present trends persist. The implementation of available and future technology can regenerate or at least retard the rapid destruction of desirable aquatic habitat.

Technology available at present which will sustain or enhance aquatic life is as follows:

1. All lakes in Florida should be allowed to fluctuate on a wide range to simulate natural conditions thereby stimulating game fish production. In many cases, the lakes may need to be severly drawn down to set back the eutrophication process.
2. More efficient nutrient removal and utilization of industrial, domestic and agricultural effluent, eg., photosynthetic sewage treatment, spray irrigation sewage treatment.
3. Stringent zoning legislation should be enacted to protect all flood plains.
4. All dredging and filling, except for enhancement for the aquatic environment, should cease.
5. Stricter legislation should be enacted to reduce and control the pesticides and herbicides used in the environment.
6. To facilitate nutrient removal and full utilization of the resources, increased fresh water fishing harvest techniques of certain species should be employed.
7. Also, to facilitate nutrient removal, there should be a development of a mechanical aquatic weed harvester which can transform aquatic weeds to animal food supplements.

8. Investigate desirable natural and exotic fishes which exhibit superior short-term qualities and can live in an adverse aquatic environment.
9. Screen commercially harvestable native and exotic species which exhibit a dual purpose of removing nutrients and food production.
10. Applicable short-term management techniques may involve selected rotenone treatments and total renovation of the fish population in some regions.
11. Investigate various organisms, native and exotic, which are capable of biologically controlling problem aquatic plants.

Cultural practices which either contribute nutrient materials to the ecosystem or accelerate detrition by enduced recirculation of nutrients within the system, result in environmental changes which persist after the practices have been discontinued.

Those intent on grandiose projects to mechanically and chemically remodel our environment have too often found that while they are experts with structural steel, concrete, ditches, dams, chemical formulas and waterways, they are not trained in the application of ecological principles. The tragedy is in the fact that much of the aquatic and environmental destruction could have been avoided in the original planning.

By now you are probably asking yourself what all of this has to do with the future of fresh water fisheries in Florida. We say it has everything to do with it. We cannot continue to develop high quality sport fishing in waters that are becoming unfit for the survival of fishes. It is interesting to note that a society such as ours, where man can remove himself to a certain degree from the influences of nature, continues to ignore the fact that we are changing environment to such an extent that some of the lower animals cannot survive. If all this sounds a little dramatic and removed from your actual situation, don't you believe it! Nothing is more important in our existence than reversing exploitation and devastation of our environment. If it continues at the present pace, we won't have time to worry about whether the bass are biting in Lake Jackson or how big the specks are in Lake Okeechobee--we will be too busy trying to survive.

INVESTIGATION: OBSERVATION OF SOIL MICRO-ORGANISMS

BACKGROUND: When examining life in the soil or the soil itself, we generally remove soil samples. There are times when this is done for investigative purposes. It would also be of benefit to sample organic relationships in the soil. We can do this by using the buried-slide technique. The buried-slide technique will give some idea of soil population and its relationship to environment. This may answer questions about algal colonies, fungi formation, and production of spores in soil that have been produced on agar but will this occur in the soil? The technique can be performed at school or home site.

PURPOSE: Microscopic observation of soil micro-organisms.

MATERIALS: Several outside locations at different depths; glass slide; rose bengal stain; burner; water bath for staining.

PROCEDURE:

1. Select several outdoor sites in different areas around school or home site. (Try a shaded area, a sheltered area, and an open field area).
2. As you place your slides in the ground, mark them so you will find the slides after the incubation period.
3. Place clean slides into the soil at different depths, allow slides to remain in soil for at least 7 days (this investigation can be done in the classroom with a mixture of 466 g. of water, 1 g. cornflakes, and 200 g. soil kept at 28° for 7 days. A similar investigation with water could be done by suspending the slide in water near school or home site at different levels for at least seven days).
4. Remove the slide from the ground being careful not to disturb one side of the slide. This can be done by tilting the slide about 45° before removing from soil so that one side is not distrubed.

5. Flame fix the slide. This is done by passing the slide through the flame.

6. Now that the slide is flame fixed, set up a water bath for the slide. Place the slide on top of the water bath. Drop the stain on the slide and continue to add stain not allowing the slide to dry. Stain for about 8 minutes, keeping water bath operating. Do not stain any less than 5 minutes or more than 10 minutes.

7. Remove the excess stain by rinsing the slide under water for a few minutes. Allow slide to air dry and observe the results under a hand lens or low power microscope.

INVESTIGATION: MICRO-ORGANISMS IN THE SOIL

BACKGROUND: The number of micro-organisms found in the soil depends on the type of soil. A rich loam suitable for gardening may contain as many as 5,000,000,000 bacteria per grain. Most of this micro-life exists near the surface, where organic matter accumulates. Micro-organisms can modify and convert this organic matter into their own nutrients. Relatively fewer micro-organisms are present at subsoil level. Organic decay at the subsoil level will depend on conditions of moisture and drainage.

PURPOSE: To determine the presence of micro-organisms in the soil.

MATERIALS:

Millipore kit
Soil sample
Dilution tubes

PROCEDURE:

1. Sterilize six small glass dilution tubes; the sterile apparatus, funnel, and filter support.

2. Fill one of the dilution tubes 1/3 full of the soil sample, add a few millileters of sterile dechlorinated water. Fill remaining tubes with sterile dechlorinated water.

3. Using a flame-sterilized dilution loop, transfer one loopful of water from the top of the sample tube to the next tube (consider this tube as number 2). Swirl the tube to mix.

4. Be sure to flame the loop before each use; transfer a loopful from tube number 2 to tube number 3 and swirl to mix; transfer a loopful from tube number 3 to tube number 4 and swirl to mix and so forth until all of the tubes have been inoculated with progressively decreasing concentrations of micro-organisms.

5. Pour contents of tube number 6 (tube with the highest dilution) into the sterile funnel and filter the contents through the first test filter.

6. Prepare a petri dish and pad with total count medium (yellow). Transfer the test filter to the pad, and close the dish.

7. Repeat above process with the contents of dilution tubes 4, 3, and 2.

8. Incubate them for 48 hours.

9. Write your observed results.

INVESTIGATION: TERRESTRIAL

PURPOSE: Australian pine needle effects on plant life.

MATERIALS: Australian pine needles; alcohol; lanolin paste (drug store item); seeds; seedlings; styrofoam; aluminum can or flower pots for seeds and seedlings.

PROCEDURE:

1. Collect Australian pine needles and place them in a 250 m. beaker.
2. Cover needles with alcohol. Cover the beaker. Allow this mixture to set for 2 or 3 weeks. Do not allow the alcohol to evaporate during this time.
3. After 2 or 3 weeks allow alcohol to evaporate from the solution, thereby leaving a residue in the bottom of the beaker.
4. Mix residue with the lanolin paste. Spread paste on the seeds and on the several leaves of the seedlings. Be sure to plant some seed and some seedlings that have not been treated with the lanolin paste. (This will be your control).
5. After planting, keep daily records of the plants for a period of three to four weeks.

Questions:

1. Did any of the seeds with pine-lanolin paste survive?
2. Why do you think this happened?

INVESTIGATION: SOIL AS AN ECOSYSTEM

BACKGROUND: Pages 234 - 247 BSCS Green Version

INVESTIGATIONS:

- pH
- Moisture Content
- Water Capacity
- Soil Composition
- Soil Algae
- Terrestial (Organic Centent)
- Investigation: Soil Life
- Observation of Soil-Micro-organisms
- Nitrogen Content (Hach kit)
- Millipore

The preliminary part of the soil algae investigation may be completed prior to the background study to allow sufficient time for incubation. The remaining investigations on pH, moisture content, water capacity, and soil composition may be integrated at the teacher's discretion, throughout the background study. The Soil Ecosystem Unit may then be culminated with the completion of the soil algae investigation. The importance of the chemical composition of the soil algae medium may then be illustrated and emphasized.

The above investigations may be also included in a transect study.

INVESTIGATION: MOISTURE CONTENT OF SOIL

BACKGROUND: Water content is defined as the amount of water in the soil at a particular time. It is important to know the amount of water in a soil that is available to the plant. In order to find out what is available we can measure the moisture content.

PURPOSE: To measure moisture content in the soil.

MATERIALS:

100 gram soil sample
Filter paper
Aluminum soda can

PROCEDURE:

1. Weigh a dry, empty aluminum can. Record weight ____________

2. Collect soil sample and weigh in the aluminum can. Record the weight ____________

3. Dry soil by using a Bunsen burner. Heat the soil for several minutes over an hour's time. Do this for two days. Cover the can over-night so that no moisture will get into it. (If drying oven is available, place soil in oven for 24 hours.) Record weight ____________

4. Weigh the soil after 48 hours. Record weight ____________ The weight loss will be the water loss.

5. Subtract the weight of the 48 hour sample from the original sample. (#2 - #4) (Example 60 g of soil before drying; after 48 hours, the soil weighed 50 g, then the total weight of water sample is 60 - 50, or 10 g.

6. Calculate the per cent moisture by using the formula:

$$\% \text{ moisture} = \frac{\text{Loss of weight due to drying}}{\text{Weight of dried soil}} \times 100$$

INVESTIGATION: WATER-HOLDING CAPACITY OF SOIL

PURPOSE: To measure water-holding capacity of the soil.

MATERIALS:

Dried soil from the moisture content experiment
Aluminum can
Graduated cylinder
(100 grams of dried soil if other soil is not available)

PROCEDURE:

1. Put a small hole in the bottom of the aluminum can. Now place 100 g of soil in the aluminum can.
2. Place can in a pan or beaker of water overnight to maintain moisture.
3. The next day raise aluminum can out of water and place a piece of filter paper on the bottom. Allow to drain 30 minutes.
4. Wipe the surface dry and weigh the unit.
5. Moisture-holding capacity is calculated as follows:

Per cent moisture-holding capacity =

$$\frac{\text{Gain in weight after immersion in water}}{\text{Weight of dried soil prior to immersion in water}} \times 100$$

NOTE: The gain is computed by subtracting the combined weight of the can and dry soil from that of the can and wet soil.

OPTIONAL: Acidity and alkalinity can be tested by placing a drop of distilled water on a sample of dried soil. Use litmus paper to check acid or alkaline conditions of soils.

INVESTIGATION: ORGANIC MATTER IN SOIL

PURPOSE: To determine organic matter in soil sample.

MATERIALS:

Test tube
2% solution of NaOH
Cup of soil
Graduate cylinder

PROCEDURE:

1. Place 2 cubic centimeters of soil sample into a test tube.

2. Add 6 ml of 2% NaOH to the test tube. Shake tube for 2 or 3 minutes.

3. Let the mixture stand for 48 hours. Label and cover test tubes.

4. Observe the color of the solution above the soil at the bottom of the test tube. Use the following key:

Jet black liquid - high organic matter present.

Dark brown-black - medium organic matter present.

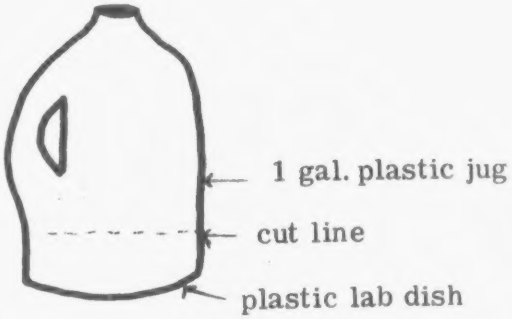
Any light shade - low organic matter present.

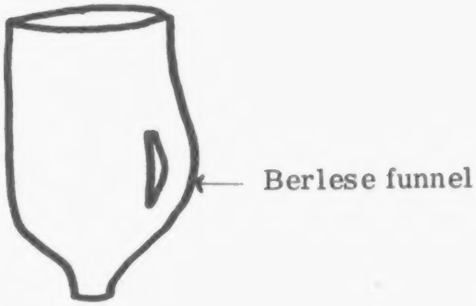
INVESTIGATION: LIFE FORMS IN SOIL

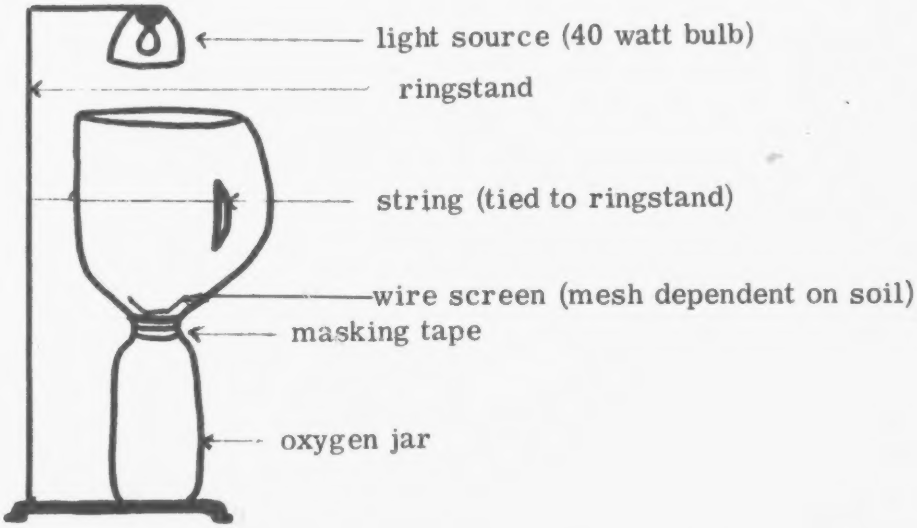
BACKGROUND: In order to perform this experiment on a terrestrial area it will be necessary to construct and set up a Berlese funnel and apparatus, if one is not available. This may be done by using the following diagram.

MATERIALS:

- Light source
- 1 gallon plastic jug
- Scissors
- Gas jar
- Masking tape


1 gal. plastic jug
cut line
plastic lab dish


Berlese funnel


light source (40 watt bulb)
ringstand
string (tied to ringstand)
wire screen (mesh dependent on soil)
masking tape
oxygen jar

Berlese funnel and apparatus

INVESTIGATION: SOIL LIFE

PURPOSE: To observe life forms in soil.

MATERIALS: Berlese funnel and apparatus; oxygen jar; 250 ml soil sample; light source (40 watt light bulb); beaker.

PROCEDURE:

1. Construct Berlese funnel and set it up on the apparatus.
2. Collect a soil sample using a can.
3. Weigh 250 ml beaker and fill with soil sample.
4. Weigh the gas jar. Record weight ________________.
5. Weigh beaker and sample. Record weight ________________.
6. Place soil in Berlese funnel and turn on light source.
7. Observe results with hand lens orbinocular microscope and identify life forms.
8. Weigh total life mass in gas jar.
9. Weigh difference between total life mass and gas jar. This is the weight of the life forms. Record weight ________________.

POPULATION & COMMUNITIES

COMMUNITY CHANGES IN PONDS AND LAKES

The number and kinds of plants and animals making up a pond or lake community changes continuously. These orderly and progressive changes are called "succession." Some of the changes are rapid, others come slowly. Often it takes hundreds of years for the succession of life in a pond to be completed. Various stages of maturation can often be seen in the different ponds in one region. In its young stage, organic matter from pioneer plants and animals and from debris has just begun to accumulate in the pond. In time, seeds of a few emergent plants are carried to the pond by the wind, water, or pond-visiting animals, and plants begin to line the shore. Then small fishes, snails, mussels, caddisflies, mayflies, and dragonflies find sufficient food to live in the pond. Some arrive as eggs carried on the feet of birds or other pond visitors. Adult insects may fly from pond to pond; frogs, turtles, mice, and other large animals travel over land.

Eventually pondweeds become abundant on the bottom, and the emergent shore plants grow farther out into the pond. All contribute more and more organic matter to the bottom as they die and decay. And, as the plant population changes in character, the kinds of fishes, insects, and other animals also change.

Finally emergent vegetation extends all the way across the pond, which now may be called a marsh. (Marshes may also originate in other ways.) Bullheads, salamanders, frogs, and turtles are the dominant large animals; worms live in the thick bottom mud, and many aquatic insects are found in the shallow, weedy waters. Land plants close in around the shore, growing in the rich humus. The filling continues until what was once a pond becomes either a grassy prairie or a forest. This is the stabilized or climax stage.

Changes in the community also occur from day to night and from one season to the next. Many animals stay in burrows or hide in the dense plant growth by day. At night they emerge and move about in search of food. Some planktonic crustaceans float to the surface at night, then return to the depths during the day. After a quiet winter, with the pond often beneath ice, life flourishes again with spring warming. Plants bloom, immature insects molt and take off in swarming flights of adults, fishes spawn, and frogs and turtles emerge from hibernation. Activity continues throughout the summer, then subsides in autumn as winter approaches and the temperature of the water drops.

WHAT WILL BE STUDIED AT THE SITE

Terrestrial site (home study or school site-individual)

1. Ground temperature (1 meter, 2 meter height)
2. Wind speed, wind direction
3. Cover or shelter (trees, building)
4. Cloud cover, cloud type
5. Rainfall
6. Soil type
7. Ground cover (grass, pine needles)
8. pH (generally, sand front or ocean side, pH 8; other, or leeward, pH 4.5)
9. Phytometers
10. Air humidity
11. Population counts
12. Photographic record
13. Transects and quadrats

SOIL DATA

Date of Sample													
Time (AM, PM)													
Air Temperature													
Cloud Cover (%)													
Wind Direction													
Soil Temperature													
Alga Count													
Water - Holding Capacity													
Water Moisture Content													
Phytometer													
Turbidity													
Acidity/Alkalinity													
Organic Matter													

DESCRIBING A COMMUNITY
(QUADRAT CONSTRUCTION)

MATERIALS: 1 meter stick, 4, 12" wooden stakes, 25 meters of a heavy cord such as chalkline, hammer, 4 thumbtacks.

In making population samples, one of the more effective methods involves employing quadrats. A quadrat may be defined as an area whose adjacent sides are at right angles to each other (these sides may be of any workable dimension). A convenient size for a quadrat is 1 m^2.

In laying out the quadrat, a random base point, A, is established and a stake is driven into the ground at that point. Points B, C, and D are then located by the obvious method of laying out a square whose sides are each 1 m in length. Having established all four points by means of wooden stakes, a cord is run between adjacent stakes and secured to the stakes by thumbtacks. Strings AB & CD are marked off in 10 cm sections; a cord is tied at the first mark on AB and the other end tied to the corresponding point on CD; this procedure is repeated on each of the marks until ten cords are attached to cords AB & CD. Cords AD & BC are marked in a similar manner and similar cords attached. If this procedure is followed, a quadrat of 100 cm^2 squares, will result.

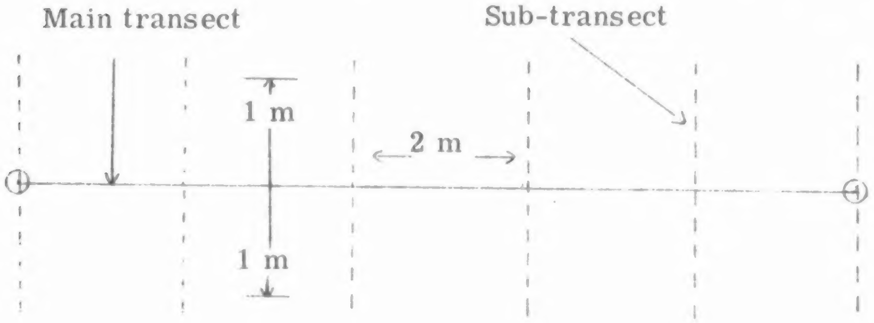
The horizontal rows are labeled alphabetically, while the vertical rows are numbered; thus a given square may be identified by row and number, such as C - 4, designating the square 3 rows up from the bottom and four rows in from the left.

DESCRIBING A COMMUNITY
(THE LINE TRANSECT)

A method of random sampling in cases where the quadrat method is not feasible or desired, is that known as the line transect. A transect might be described as a cord of any convenient length (usually, a minimum of 10 m) stretched between two points (stakes) see Fig 1 and along which the investigator takes his random samples.

The transect is most useful when sampling from one community into another (ecotone) such as from grassland into forest, or in areas of changing diversity and productivity. The transect is also used when a qualitative sampling, as opposed to a quantitative sampling, of an area is desired.

After establishing the transect, the investigator moves along the cord, sampling every 2 m along the sub-transects (1m) on either side of the main transect. Having taken samples from all areas of 20 m^2 (the length of the main transect, 10 m, multiplied by the length of the sub-transects, 1 m to the right and left of the main transect), the results may be expressed as numbers per m^2 or per unit, 20 m^2.



A Line Transect

INVESTIGATION: COMMUNITY ATTITUDES SURVEY

BACKGROUND: Man's sudden awareness of the pollution problem certainly must rank beside his accomplishments in the fields of cell biology and technology. A vogage to the moon fulfilled one of man's life-long dreams but this same high powered technology has periled any future dreams and ultimately man's survival on this planet. This survival depends a great deal upon the consumer's becoming aware of the problem and the solution. In this exercise you will have an opportunity to determine how much the average citizen knows about the nature of some of the pollution problems and how willing he is to do something about it.

OBJECTIVE: To determine the awareness of the citizenery of your community to the pollution problem and to determine their willingness to play a role in some solutions to the problem.

PROCEDURE: The basic tool of this exercise is the questionnaire. Each student should survey their neighborhood, recording the answers as to adults (21 years and above) or young people (20 or below). This data should be compiled by class on charts on the board.

RESULTS: From the totals on the board compute the following:

Compute the percentages yes - no - maybe for each question according to the two divisions.

$$\frac{\text{Total \# answering(yes)(no) or (maybe)}}{\text{Total \# answering the questions}} \times 100$$

From a study of the percentages try to determine any clear cut differences of opinion between the two groups on any or all of the questions. If any exist, state them.

DISCUSSION: If any differences of opinion occur between the groups surveyed, attempt to explain why the difference may exist.

CONCLUSION: From your analysis of the data and your discussion, attempt to make an overall statement of the results of your survey.

ECOLOGY SURVEY

"If consumers don't consume it, producers won't produce it."

	YES	NO	MAYBE
1. Would you be willing to purchase returnable soft drinks, beer, etc. to the exclusion of no deposit-no return? (plastic and glass)	____	____	____
2. Would you be willing to limit your purchase to "no phosphate" detergents until an acceptable substitute is found?	____	____	____
3. Would you be willing to vote for bond issues to construct efficient sewage disposal systems? (This means an increase in taxes!)	____	____	____
4. Would you like to see more of our "natural wilderness" area set aside just for the sake of preservation for future generations?	____	____	____
5. Would you be willing to accept a lower quality fruit and vegetable at your local supermarker at the expense of a marked reduction in the use of pesticides by the growers?	____	____	____
6. Would you be willing to sort paper wastes each week (newspaper, magazines, and others) in order to facilitate ease of pick up for recycling? (This means tying up the bundles)	____	____	____
7. Would you be willing to pay higher prices for products so that industries can install pollution control devices which would eliminate or reduce pollution of air, land, and water? (Do you really think the industries will absorb the costs without passing it to you--the consumer?)	____	____	____

	YES	NO	MAYBE
8. Would you be willing to reduce or eliminate as many of your paper products as possible by doing the following?			
A. Use sponge or cloth instead of paper towels?	______	______	______
B. Use washable diapers not disposable?	______	______	______
C. Use washable glasses and plates instead of disposable ones?	______	______	______
D. Use cloth napkins instead of paper?	______	______	______
9. DO NOT litter! This is the easiest pollution to stop.	______	______	______

INVESTIGATION: A STUDY OF FLORA AND FAUNA OF A COMMUNITY

BACKGROUND: The character of any community is primarily determined by the plant life present. Since plants are the dominant features they are more often than not used to determine the name of the community. More subtle is the effect these dominant plants play in determining the types of animals present. Since they serve as a food source, only those animals that feed on them are likely to be present. In this study the students are given methods to describe the plant community. With a knowledge of the plant life present, perhaps a better understanding of the animals will be gained.

PURPOSE: To determine the type of community by analyzing the nature of the plant life.

MATERIALS:

String (10 meters)
2 meter sticks per team of 4 students
Pencil
Paper

PROCEDURE: A list-count-quadrat method will be used here. Students should be divided into teams of 4 with each being assigned one of the following tasks:

2 students - plant counters
1 student - plant recorder
1 student - animal recorder

It is not imperative to know all the plant species but would enhance the value of the study if the major plants are known. Those of seemingly lesser importance can be given common names agreeable by all. For that matter, all the plants can be given common names. However, the students should either be familiar with them, or have available a handout with diagrams, so that they can all give the same plant the same name.

A starting point and finishing point should be given to each team who then proceed as follows:

1. At the starting point, make a square meter with the meter sticks. The plant counters should count all the plants of each species within the square and give the information to the plant recorder. The animal recorder's sole responsibility is to observe the plot for all animals present or signs of animals (tracks, feces, holes, etc.) and record the data (numbers aren't necessary for animals unless desired).

2. Upon completion of plot or station #1, the group should proceed in a straight line to station #2 which is 10 meters from the starting point. At station #2, the square meter is again established and the count of all plants and of the animals present.

3. The procedure should be repeated until at least 10 plots have been completed. (The going will be more difficult in dense undergrowth than an open field!)

RESULTS:

1. Relative density - a calculation of the percentage of the total plant count a certain species is. From the list count the students should:

a. Count the total number of all species.

b. Count the total of each species.

With these data use the following formula to calculate the relative density of each species:

$$RD = \frac{\text{total number of species X}}{\text{total number of all species}} \times 100$$

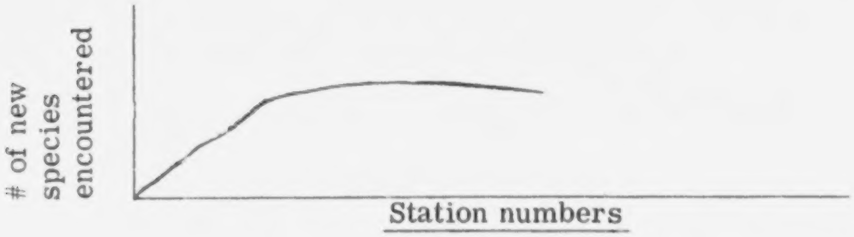
2. Frequency - density of a species in a given plot. If the number is low, the species may be one that occurs in patches. If the number is high, the species may be one that is prevalent in the study area. Use the plot by plot data according to the following formula:

$$\text{Frequency} = \frac{\text{\# of plots in which species X occurs}}{\text{Total \# of plots}} \times 100$$

The students should calculate the frequency of each species, placing the results in a chart beginning with the highest and ending with the lowest frequency.

3. The species area curve is an anlaysis of the sample size. It indicates whether or not the sample size was large enough to adequately describe the community under study. The following steps should be followed:

a. Using a piece of graph paper, prepare on the horizontal axis the number of plots in the sample. On the vertical axis, prepare a scale that includes the total number of species in the study. At station #1, the total number of different species encountered should be graphed. At station #2, a count should be made of the number of new species encountered, ie - the number of species that are different from station #1. The procedure is repeated for station #3. All the new species encountered in station #3 that are different from station #1 and station #2 are graphed. Repeat the procedure until all stations have been graphed. Interpretation is based on the type of curve obtained. If the sample size is large enough and therefore valid, the curve should level off as below:



If the sample size is not large enough, new species will still be encountered, therefore causing a continued slope in the line.

4. Analysis of the animal data. The list of animals observed directly or indirectly, should be listed on the board. From this list, the student should attempt to diagram the probable food web for the community. Reference texts should be made available so that the source of food for the animals can be determined by the student.

DISCUSSION:

1. From your relative density calculate which of the plants seem to be the most frequent?

2. What are some of the rarer plants of the community?

3. Based on your data, what do you think this type of community should be named?

4. Which species seems to be found everywhere in the community? What was the frequency?

5. Which species seems to be found in only one spot of a few places? What was the frequency?

6. According to your species area curve, was your sample large enough? How do you know?

7. From your animal data, do you have any 4th order consumers? Calculate the percentage of 1st, 2nd, 3rd, and 4th order consumers in the area. Explain the results.

CONCLUSION: Make a general statement based on your results on each of the following:

A. The name you gave to the community.

B. The frequency of plants in the community.

C. The relative density of plants in the community.

D. The sample size of your study.

E. The animals of the community.

REFERENCES:

Kormondy, Edward J., *Concepts of Ecology*, Prentice-Hall, Inc., Englewood Cliffs, New Jersey, 1969.

Smith, Robert Leo, Ecology and Field Biology, Harper and Row, New York and London, 1966.

INVESTIGATION: REDESIGN OF AN EXISTING CITY

OBJECTIVE: To provide practical experiences in socio-economic problems involved in reconstruction.

MATERIALS:

Large city maps
Aerial photo of city
Poster
Paper
City directory
Zoning manual (city and county)

PROCEDURE:

1. Maps, photos and zoning ordinances can be obtained from city and county government.

2. Explain project to class. Provide few restrictions but do give some buildings and other structures which may not be changed by the students to introduce bias to the students. Practicality and expense are not to be considered at this time.

3. Divide the class into groups and let each group devise a rough plan. Entire class should elect one chairman.

4. Chairman moderates a class discussion on the merits of each group's plan. This may get a little rough but best results will come from this type of settlement.

5. After general plan is agreed upon by class have chairman divide class into groups to gather information from sources listed or other sources.

City Mayor
City Council
City Planning Commission
City Zoning Commission
City Utilities and Water
Chamber of Commerce
County offices similar to those in the city

This information will contain data such as census, plans for future development, industrial expansion, zoning ordinances, and utility usage. All of this will be necessary for final city plan.

6. Assign groups to various city services such as fire protection, police protection, government, and utilities in order to plan growth and changes with the "new plan".

7. At this point the class will have decided upon what general layout they want and what type of dwelling they would use in the new city. Invite a guest speaker to class to talk with the students. This speaker should be capable and knowledgeable in socio-economic problems.

8. Review plan in class. Watch for changes as a result of guest speaker.

9. Draw up map and final report.

INVESTIGATION: MICROSUCCESSION IN THE ROTTEN LOG

Succession from the standing dead tree-to the fallen log-to the humus of the forest floor may take many years for a given tree; however, by examining several logs of the same species in the same community at various stages of decay, the succession may be observed in a matter of one or two hours. The stages or phases may be described as four distinct conditions of the logs: Phase A would be the standing dead tree; Phase B, the newly fallen log; Phase C, the log still retains the hard exterior while rotting inside; Phase D, the completely rotted log.

Questions to be considered, or hallmarks to look for are as follows:

Phase A Is the bark still on the tree? Is it still firmly attached or not? What invertebrates are observable in connection with the log; that is, are they under the bark? are they in the wood? Is the wood dry under the bark? Have any vertebrates established nests in the tree? The student should make a record of all animals found, both as the species and numbers.

Phase B Is there still bark on the tree: If so, does it lift off easily? Is the wood hard or soft? wet or dry? What invertebrates are found under the bark or in the wood? Are any vertebrates nesting in it at this state? (A hand ax might be a handy peice of equipment to accompany this observation.)

Phase C After breaking the "shell" of hard exterior and examining the contents by raking through the fragmentized portion of the log, are there lizards, salamanders, or snakes present? Are there identifiable eggs present? What invertebrates are present? Is there any indication that the log may be a part of the small mammal "highway system"?

Phase D Having raked through the rotted wood, as in Phase C, is there more or less moisture present than in the preceding stages? What types of animals are now making the log their home?

Conclusions: Which stage contained the most diversified animal community? By species, which stage had the most individuals? Can the chemical differences between the first and last phases be described? If so, how? which are the most outstanding physical differences between the first and last phases?

Equipment: Hand cultivator (of the type used by gardeners) for purposes of raking through the detritus, belt ax or geologists pick for investigating Phase C, recording book, pencil, Scout-type knife.

MICROSUCCESSION IN ROTTEN LOG

DATA SHEET DATE ____________________

QUADRAT NO. ________________ AIR TEMP. ________________

LOG SPECIES ________________ SOIL TEMP. ________________

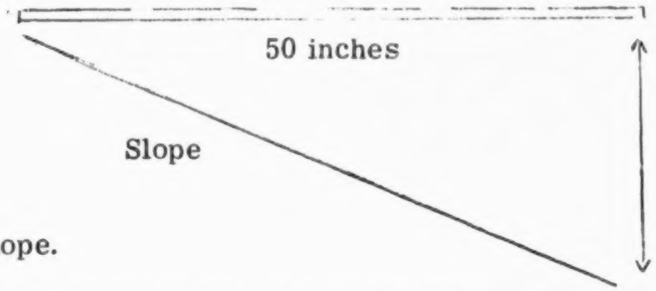
PHASE A General description of log		PHASE B General description of log	
Species	No.	Species	No.

INVESTIGATION: SCOUR EROSION ON CANAL BANKS

PURPOSE: To show affects of scour erosion.

MATERIALS: Level (or glass tube half full of water) ; 50 inch stick; yard stick.

PROCEDURE: Select slope to be measured. Place end of 50 inch stick at top of slope. Raise other end until level-use level to be sure. Measure distance from free end of stick to ground.



height x 2 = % slope.

By repeating this procedure at regular intervals through the semester a chart showing the change in slope and rate of change can be drawn. Using the graph example below, plot a semester graph of a slope at your site. Different ground covers on various slopes will show effectiveness of cover in slowing erosion.

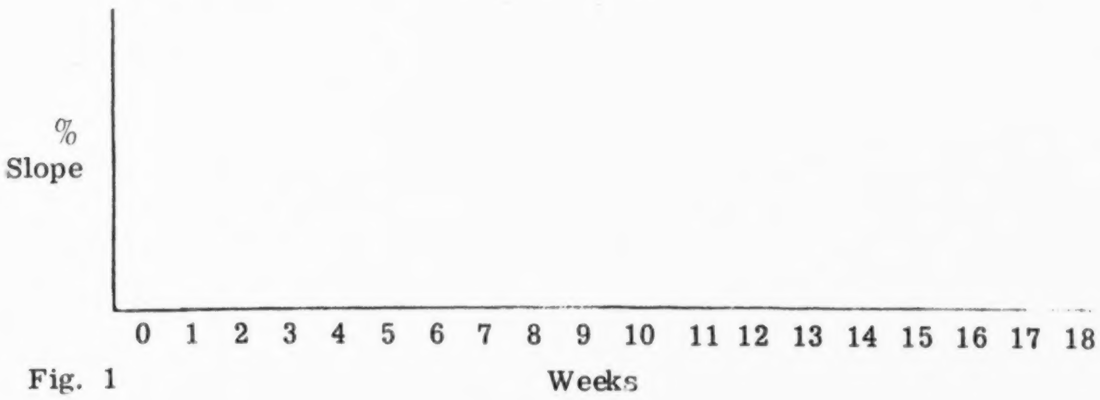


Fig. 1

INVESTIGATION: TERRESTIAL OR AQUATIC LIFE

OBJECTIVE: To do a one specie population study at a series of sites.

BACKGROUND: It is possible through the study of one specie in a community to become more aware of the interrelationships of the community. There can not be a preset procedure as the choice of specie and community may cause wide variation.

This is an investigation that is most effectively run in teams. In case data must be collected on days one student would be absent, the investigation would continue uninterrupted.

The students should choose a specie and gather reading background on the specie to be studied while they are gathering data in the field. The student should choose several environmental sites and place traps to collect live species. These traps should be checked daily at a specific time. If any live specimens are caught, the students should tag the specimens before releasing them. Tagging methods can be obtained from various field biology books. Collect data on specimen and release captured specie. Careful records should be kept on the investigations.

Students should map areas from which samples are taken. This may help establish a clearer understanding of species interrelationship to its community.

The data should register number of specie, date of the the catch, location, length, color, sex, tagging used, time of find, time of release; if the specie is recaptured repeat the same records, and release. Finally, a graph of the population should be made covering the period of study. Conclusions should be drawn from data collected.

INVESTIGATION: SUCCESSION IN A MICROCOSM

BACKGROUND: Most communities of plants and animals tend to change. A group of organisms is replaced by a second: the second may be replaced by a third. Each new organism that gains supremacy alters the environment and provides conditions that favor other species. Thus, populations may change from hour to hour and day to day. Some successions, such as changing organisms in a temporary rain puddle, occur on a very small scale. Such a succession is usually over in a matter of days. Other successions, such as a change from bare ground to mature forest, may take a century or more.

MATERIALS:

5 jars to collect pond water
Cultures of pond water
Microscope
Hydrion paper
Pipette
Centigrade thermometer
Slide
Cover glass
Light meter

PROCEDURE: Select a stream, pond, or lake from which you can collect your water cultures. Fill five quart jars three-fourths full of water containing some plant material. Some cultures should contain floating vegetation such as duckweed, others should contain algae only, and one culture should contain some submerged vegetation and dead plant material. This will create a different environment for each culture.

Place the cultures in an area of the laboratory where each will receive the same amount of light. If you have a light meter, check the light at various times of the day. (a) What is the variation in foot candles of light available to the cultures? (b) Why is light important in the study?

Using a centigrade thermometer, check the temperature of the culture each day that you examine the water. (c) Why is the temperature an important factor in succession in the jar?

Using Hydrion paper, check the pH of the medium each day of the examination and draw a line graph of the changes. (d) How is the pH of the medium related to succession? (e) What factor is responsible for the change in pH?

Examination of the cultures should be made on alternate days for a period of three weeks. Since organisms occur at all levels of the culture remove a drop from the surface, a drop from approximately 4 cm from the surface and one from the bottom for study. Study each drop of water under the low power of the microscope and determine what organisms are present.

In the five drops taken from the surface, count the number of each species present. Determine the average number of each species in the five drops.

Set up a chart on which to record these averages. The chart should include such groups as green protists, nongreen protists, rotifers, crustaceans, annelids, gastrotrichs, and others, as you find them.

(f) What type of organisms did you find on the first day of examination?

(g) How long did these remain in the dominant organisms?

(h) Describe any changes that occur in the water.

(i) What caused the changes in the water?

(j) What organisms assumed dominance after the water became turbid?

(k) Explain why these organisms became dominant.

(l) Explain why the cultures became clear at the end of the experiment.

(m) What organisms followed Colpoda and Colpodium in the succession?

(n) What other organisms occurred in the succession?

(o) When did the rotifers and gastrotrichs appear?

(p) What factors govern the succession in the water cultures?

INVESTIGATION: OBSERVATION OF FLORA AND FAUNA

PURPOSE: To study the principles of ecology by observing the flora and fauna found around the school.

MATERIALS: An outline map of the school grounds; funnels; beakers; 100 ml graduate; ring stands and rings; microscope slides; cover glasses; methylene blue; absorbent cotton.

PROCEDURE:

1. Walk around the area under investigation recording the numbers and location of plants on the outline map. Use symbols T for trees, S for shurbs, P for non-woody plants, and G for grass.
2. Record any rocks that might be on the grounds by using R.
3. Record the location of the areas of any birds seen with the letter B.
4. Record any insects as I and worms as W.
5. Turn over any rocks found and record the kinds of animals found there, also record their activities. Return rock to original position.
6. Bring back to the classroom some samples of soil from various parts of the grounds and from around the roots of some clover plants if any were found. Test the water retaining properties of the soils by placing each of the samples into a funnel over a piece of absorbent cotton. The cotton will prevent the soil from being washed through. Place the funnel on the ring on the ring stand so that the water will be allowed to go through, from the time the first drop is poured into the funnel until the last is filtered through.

7. Prepare a slide of a extremely dilute solution of soil from around the roots of clover plants. Stain with methylene blue, and examine under low and high power of the microscope. Make drawings of any organisms seen.

8. This could be studied over a long period of time as a succession project.

STUDYING THE DATA AND CONCLUSIONS:

1. Have the plants and trees used as landscape been placed in their locations for any specific reasons?

2. What have you noticed about the wild-growing plants?

3. Where did you find the birds? What kinds of birds did you find? Water birds? Predatory birds?

4. Do worms and insects do anything for the soil?

5. Were the animals found under rocks, vertebrates or invertebrates? Why?

6. Did you observe any organisms in the soil water? Are they of any particular value in respect to other organisms--ecological relationship?

7. To what cycle do the organisms found in the soil from around clover roots belong?

8. Do you have part of the nitrogen cycle in operation in this exercise? Where?

9. What is the ecological relationship between the composition of the soil and the types of plant and animal life it supports?

10. Can you think of any exercises for further study?

INVESTIGATION: RANDOM COUNT

BACKGROUND: This experiment deals with the study of terrestrial population. The study can be run in a field around the school site and at a home site. The control point of this experiment should be focused around one population such as a weed, flower or insect. This will be a preparatory lab for students work with quadrats.

PURPOSE: An exercise in random sampling.

MATERIALS:

Graph paper
Chart per example
White adhesive tape (3 inch strip)
Wire coat hanger

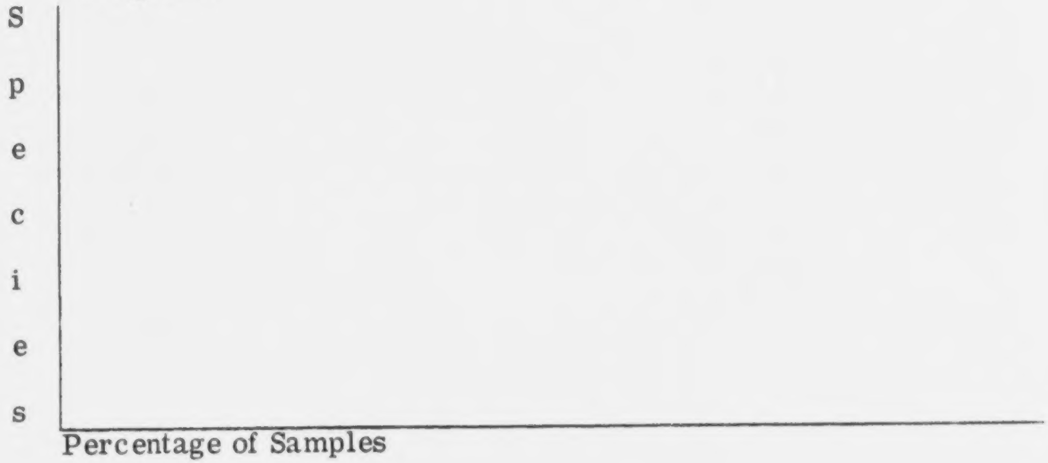
PROCEDURE:

1. Bend the coat hanger into a circular hoop.
2. Place the 3 inch adhesive strip around the wire hoop.
3. Throw the hoop on the ground and record the name of all the individual plant samples in the hoop.
4. Court the weeds and record their occurrence.
5. Repeat steps 3 and 4 until 50 samplings have been counted and recorded.
6. Add any new weeds to the above list.
7. Plot the percentage as a bar graph or histogram.

Observations:

Species	Number of Samples	Occurrence	Percentage
	1 2 3 4 5 6 7 8 9 10 11 12 13 14 15 16 17 18 19 20 21 22 23 24 25		

Histogram:



INVESTIGATION: SMALL MAMMAL POPULATIONS

BACKGROUND: Open fields usually support a relatively large population of small mammals; however, unless the observer manages to be "in the right place at the right time", the fields could, conceivably, be considered a sterile desert insofar as faunal population is concerned. Therefore, the investigator must utilize a method of sampling which does not involve the actual observation of the animal; evidence of presence is all that is required. A common evidence of the small mammal is the fecal deposit, or scat. Small mammals usually defecate in a cleared area, hence, scat boards provide a simple means of determining both population density and/or range.

Masonite or exterior grade plywood may be cut into 10 cm x 10 cm squares. These scat boards are then distributed in a uniform grid pattern throughout the habitat at 15 to 30 meter intervals. Small animals will use these boards for defecation sites, hence, leave an indication of their presence. Generally, 100 boards are used and must be visited once during every 24-hour period for three consecutive days.

To further increase the usefullness of scat boards, dyed baits may be utilized to detect movement within the population. Non-toxic dyes such as Fluorescin may be mixed with a bait such as oatmeal. By using different colored baits in various parts of the habitat, movements of small mammals may be detected.

By feeding dyed bait to a captured animal and releasing him, then observing/collecting scats, movement by an individual may be ascertained.

Questions:

1. What was the population of small mammals per acre?
2. Was there evidence of parasitic infestation?
3. Was there evidence of disease?
4. Would you say that the small mammal population was low, medium, or high?

INVESTIGATION: POPULATION STUDY OF GOPHER TORTOISES

BACKGROUND: The gopher tortoise, Goperus polyphemus, is wide spread throughout the Florida peninsula. They prefer open fields, pine scrub, or palmetto pine-flatwoods, although many of their burrows can be seen around the banks of sand piled beside canals. They dig tunnels at an angle that slopes downward from the surface for 10 to 35 feet and eventually leveling off into a large enough room for them to turn around inside. Many other animals usually share the gopher's home. One species of burrowing owl prefers the gopher's tunnel. Other animals living commerserately with the gopher are raccons, possums, gopher frog, indigo snakes and the deadly rattler.

Tortoises usually emerge only in warm winter weather and in the summer, only in the early morning and late afternoon, avoiding the noon day heat. In this exercise, students will attempt to determine the density of gophers by observing their burrows for signs of activity.

MATERIALS:
Sticks (picked up on the site)
Tape or measured string (to measure the study area)
Tags or cards (for claiming the gopher hole)

PROCEDURE:

1. Smooth the sand in front of the burrow. CAUTION: These holes house other animals as well as tortoises. Rattlesnakes will not like the intrustion. Caution should be exercised near the mouth of the burrow.

2. Place small twigs in a fence-like manner in front of the

burrow. Leave enough space between the twigs so that other inhabitants may exit and return without knocking down the fence.

3. Observe the burrow the following day. Tracks and flattened twigs indicate an active burrow and the presence of a gopher.

If there are many gopher holes and the study area is large enough, each class can be assigned a specific area to cover. Within the assigned area, each pair of students can find at least one gopher hole and by placing a sign near the hole, claim the burrow as theirs. It is important that all burrows be "claimed." Also the total area under study must be measured.

RESULTS: Density of gophers can be calculated when the totals have been placed by classes on the board.

$$\text{Density} = \frac{\text{No. of sq. meters in the study}}{\text{No. of positive gopher burrows}} = \text{area per gopher}$$

DISCUSSION:

1. If the area studied was divided among the classes, calculate the density of gophers for each class.

2. If there were differences in the densitys of gophers in the area, look carefully at the physical features of the study and speculate the possible reasons for the differences.

3. What are some of the possible food and water sources for the gopher tortoise.

CONCLUSION: Make a general statement about the gopher tortoise population density in the area of study.

REFERENCE:

Conant, Roger, *A Field Guide to Reptiles and Amphibians*, Houghton Mifflin Company, Boston, 1958.

Hollister, O.D., *Marine Science Teacher*, Melbourne High School, Melbourne, Florida.

INVESTIGATION: FLORIDA COTTONTAIL POPULATION

OBJECTIVE: To determine the population density of Florida cottontails.

BACKGROUND: A common nocturnal sight in Florida is the cottontail, Sylvilagus floridanus. These animals prefer the semi-open palmetto communities and seem to be oblivious to the presence of human dwellings near by, as indicated by the presence of droppings in yards or a bounding cottontail in the headlights of a car turning into the driveway. These shy animals play the role of primary consumers in almost all of Florida's communities and they provide healthy meals for the fox, eagle, and other carnivores.

This exercise is designed to roughly estimate the density of the cottontail in any given community. Rabbit droppings are used to determine the presence of the animal. The method described below requires that a rabbit be kept in the lab or at a student's home so that the weight of droppings can be determined in a given period of time. This becomes the standard by which the population "in the wilds" is established.

MATERIALS:
- String for measuring sample plots
- Forceps
- Balance

PROCEDURE: The area under study and the number of students involved will determine the sample plot size. If the study area is small, it is a simple matter to divide the area equally among the students. If the area is large a sample must be taken at random. Assuming the latter to be the usual case, each student could be assigned a 10 sq. meter plot for which he is given the responsibility for two tasks: (1) remove all droppings from his sample area and (2) carefully observe after 24 hours and collect all fresh droppings. These droppings must be weighed and the total recorded. It should be kept in mind that the greater the difference in sample size and study area the greater the lack of validity. Enhancement of validity can occur also if the procedure is repeated for several successive days and the average used in calculations.

Food placed in the sample area would encourage cottontails into the sample area and increase the possibilities of droppings.

RESULTS: If the total study area has been divided equally among the students the calculations are simple:

$$\text{Rabbit Population} = \frac{\text{Total wt. of droppings in study area}}{\text{Total wt. of droppings in 24 hours from standard rabbit}}$$

This gives a working idea of the number of rabbits in the study area. If the area is large and sample areas are used, the calculations are as follows:

1. $\dfrac{\text{Wt. of droppings in sample area (24 hours)}}{\text{Total wt. of droppings from standard rabbit (24 hours)}}$

This gives the number of rabbits in sample area.

2. Then to calculate the number of rabbits in the total area the proportion is:

$$\frac{\text{No. of rabbits in sample area}}{\text{Total area of sample}} \times \frac{\text{No. of rabbits in total area}}{\text{Total area of study}}$$

DISCUSSION:

1. Calculate the total number of rabbits in your study area.

2. What do you think the food source might be for the cottontails in this study? Do you think the density would be the same for all Florida communities? Why or Why not?

3. List some ways you might be able to improve the validity of this exercise.

CONCLUSION: Make a general statement of the number of cottontails in this particular statement.

INVESTIGATION: CASTING ANIMAL TRACKS

PURPOSE: To provide a method by which students will recognize individual animal tracks when encountered in the field.

MATERIALS: Forceps; plaster of Paris; poster board strips two inches wide of various lengths; india ink; small brushes; spray can of shellac or plastic.

PROCEDURE:

1. Clean track of all debris, spray track with shellac or plastic.
2. Form poster board circle around print and press into ground to create a pouring form (Fig. 1).
3. Make certain that there is at least 1" poster board above the surface.
4. Mix plaster of Paris to the thickness of pancake batter.
5. Pour the mold full of the mixture. Allow it to harden before lifting out of track (Fig. 2).
6. Clean surface of cast and coat with vaseline.
7. Form mold around cast and pour mixture until level with top of mold. If the track case is to be hung on a wall place wire loop in surface of mixture. Let harden at least two hours (Fig. 3).
8. Remove mold and separate the two layers. Wipe vaseline from cast (Fig. 4).
9. Scrape and wash cast - smooth with fine sand paper.
10. When thoroughly dry paint inside of track with india ink and label.

CASTING ANIMAL TRACKS

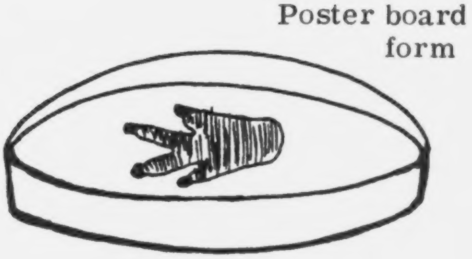

Poster board
form

Fig. 1

Pour this mold full of
plaster mix.

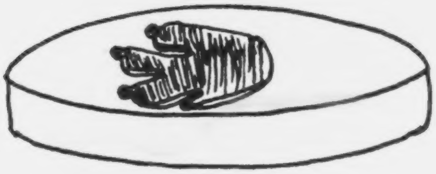


Fig. 2

Plaster Cast

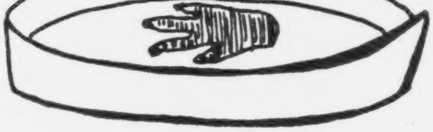


Fig. 3

Poster board mold around
first cast-coat cast with
vaseline.

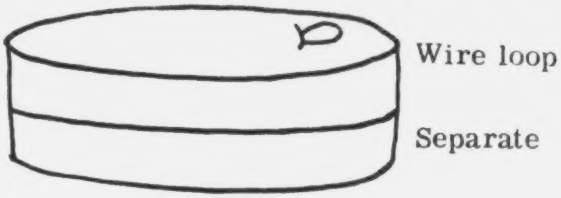

Wire loop
Separate

Fig. 4

Remove mold around
the two layers of casts and
separate at joint.

INVESTIGATION: SOIL ORGANISMS - FUNGI

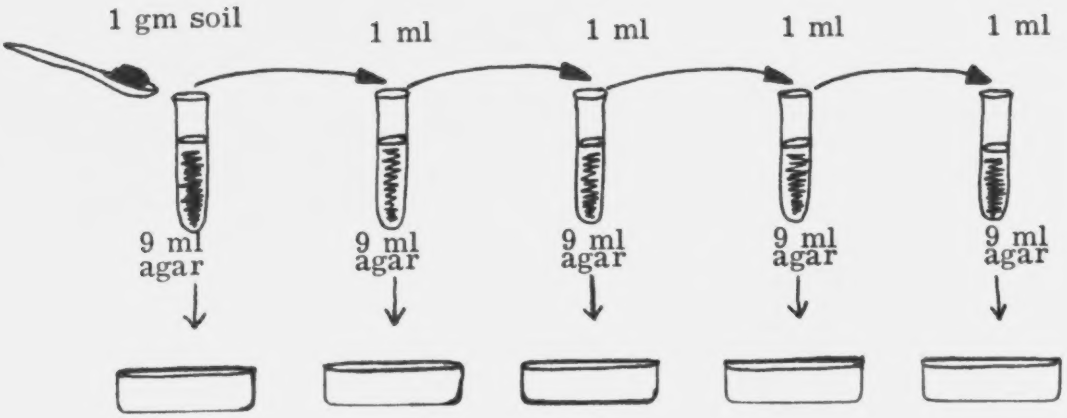
OBJECTIVE: To determine the extent and variety of fungi in Florida soils.

BACKGROUND: Fungi are important organisms in any ecosystem in that they assume the role of decomposers. Their presence can be noted as fuzzy material in soil litter or as large "orange -slice looking" bracket fungi growing on fallen or even standing trees. They also are responsible for the musty smell one encounters in the dense vegetation of a hammock. Fungi germinate from microscopic spores and produce thread-like filaments called hyphae (hypha singular). These may become extensively branched forming a dense network of mycelia. It is in this stage one notices the fuzzy appearance such as on bread, dead leaves, etc. Fungi send up readily visible fruiting bodies which produce spores. It is hoped that some soil fungi can be isolated and observed in this exercise.

MATERIALS: Sterile 1 ml or 10 ml pipette
5 tubes sterile nutrient agar (pH adjusted to 5) or soil extract agar (see soil bacteria exercise)
5 sterile petri dishes
Soil sample (1 gram)

PROCEDURE: Weigh 1 gm of soil and place into tube #1 of agar that has been cooled to 45° C. or cool enough to hold in palm of hand. Mix soil with agar by rolling the tube between the hands. Quickly transfer 1 ml of tube #1 into tube #2 that has been cooled to 45° C. Quickly pour contents of tube #1 into petri dish #1. (It is imperative that one work quickly or the agar will harden and will not pour.) Repeat the procedure for Tubes #2-5.

DILUTION


1 gm soil
1 ml
1 ml
1 ml
1 ml
9 ml agar
9 ml agar
9 ml agar
9 ml agar
9 ml agar

	Petri dish	Petri dish	Petri dish	Petri dish	Petri dish
Dilution	1/10	1/100	1/1000	1/10000	1/100000
Amt. of soil per ml.	.1 gm	.01 gm	.001 gm	.0001 gm	.00001 gm

DATA: After 7 days, observe the dishes for fungal growth. Make sketches of the growth, noting different shapes and colors of the colonies. Record the data in the following chart. You may want to use the microscope to enhance observation.

Dilution	No. of Different Kinds	Total No. of Fungi	No. per Gram
1/10			
1/100			
1/1000			
1/10,000			
1/100,000			

QUESTIONS:

1. How does the number of fungi compare with each dilution?
2. How many different fungi do your plates contain? What are the characteristics used to distinguish each kind?
3. How many of each type of fungi do you have? Does one seem to be prevalent? Why?

CONCLUSION:

Make a general statement concerning the numbers and kinds of fungi found in Florida soils.

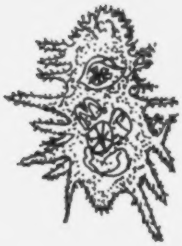
INVESTIGATION: SOIL ORGANISMS - PROTOZOANS

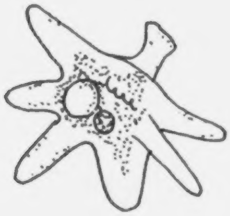
OBJECTIVE: To observe and determine the extent and variety of protozoans in Florida canal or stream soil.

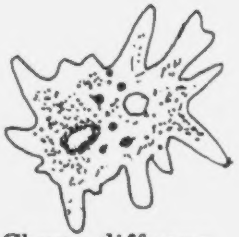
BACKGROUND: Where Florida soil is wet, life is abundant. Here among the grains of sand and particles of soil, water provides a fine environment for those organisms that may be classified as producers or consumers. Generally these organisms can be divided into groups according to locomotion. These groups are amobae, flagellates and ciliates.

Amoeba are unicellular organisms that move about with a flowing motion produced by pseudopodia. They feed by engulfing other organisms or organic debris. Flagellates are unicellular organisms with one or more long whip-like flagella which are used for locomotion. Ciliates are also unicellular but are covered with short hairs called cilia which are used for locomotion. Many other organisms may be encountered but these three groups will probable be found more often.

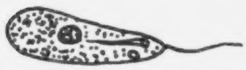
TYPICAL AMEBOID SHAPES

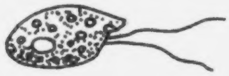

Dinamoeba horrida

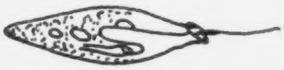

Rugpies bilzi


Chaos diffuens

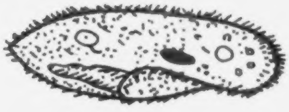
TYPICAL FLAGELLATE SHAPES

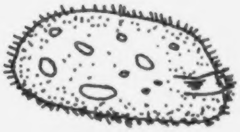

Astasia

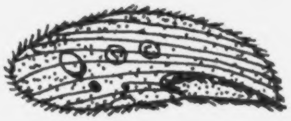

Chilomonas


Peranema

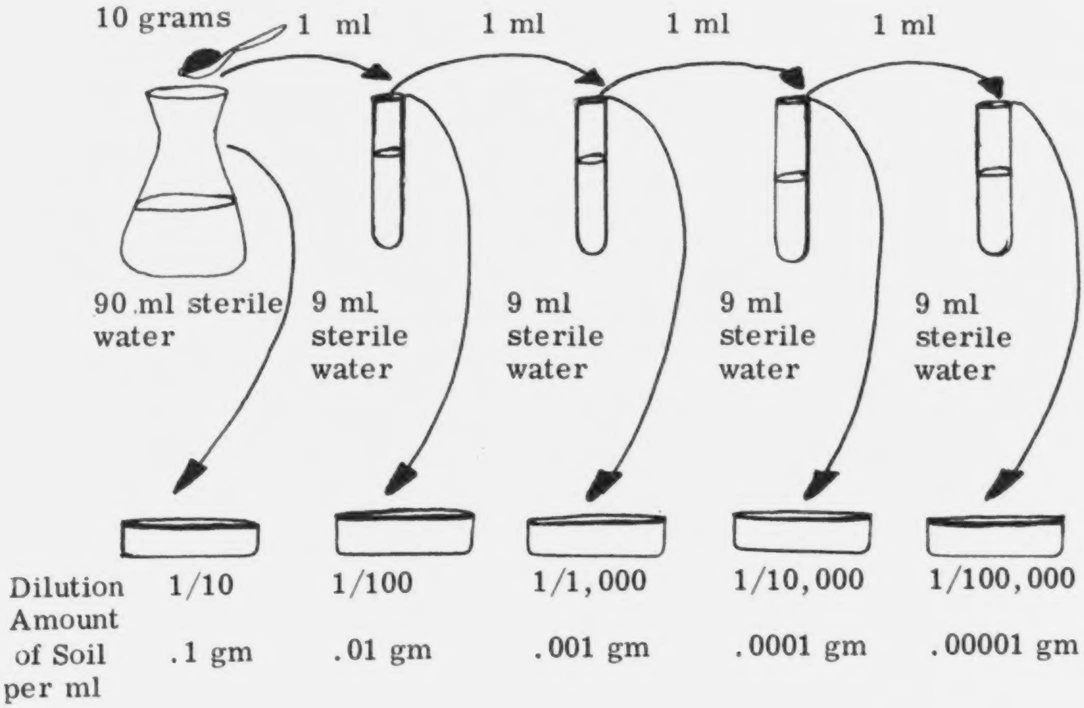
TYPICAL CILIATE SHAPES


Paramoecium


Frontonia


Loxodes

PROCEDURE: Prepare manitol soil extract agar. (see general soil bacteria exercise) Pipette 10 ml of the agar into 5 sterilized petri dishes. After the agar has solidified, add 5 ml of sterilized tap water on top the agar in each plate. Weigh out 10 grams of soil and add to 90 ml of sterile water. Proceed as diagrammed below: (use sterile pipettes at each transfer).


10 grams
1 ml
1 ml
1 ml
1 ml
90 ml sterile water
9 ml sterile water
9 ml sterile water
9 ml sterile water
9 ml sterile water
Dilution
1/10
1/100
1/1,000
1/10,000
1/100,000
Amount of Soil per ml
.1 gm
.01 gm
.001 gm
.0001 gm
.00001 gm

RESULTS: Examine the petri dishes after 7 days. Transfer, with a sterile pipette,. 1 ml of the water from the top of the agar to a clean slide. Examine under the microscope for the presence of organisms and record in the following chart.

+Present -Absent

Dilution	Amoeba	# Per Gram	Flagellates	# Per Gram	Ciliates	# Per Gram
1/10						
1/100						
1/1000						
1/10000						
1/100000						

Sketch some of the more common protozoans found.

DISCUSSION:

1. Calculate the number of each kind of organism per gram, E. G. - number of organisms counted x reciprocal of soil dilution x amount of water = number of organisms per dish.

   For this procedure be sure to cover the whole coverslip in counting the organisms. For accuracy several samples should be taken. In the formula recriprocal of soil dilution for .1 is 10, for .01 is 100, etc. The amount of water is constant - 10 ml. Perform these calculations for each dilution and each organism.

2. If soil samples are brought in from different canals, are the populations the same? If not can you explain why?

3. Which of the three groups of organisms are more numerous? Based upon the presence or absence of chlorophyll, determine how many producers and consumers you have. How do the numbers compare with each other?

CONCLUSION: Make a general statement about the numbers and kinds of protozoans found in Florida soils.

INVESTIGATION: SOIL ALGAE

OBJECTIVE: To identify and determine the quantities of algae in Florida soils.

BACKGROUND: Soil contains various forms of life and among these are algae, fungi, and bacteria. Algae are divided by color into Chlorophyta (green) Chrysophyta (gold) and Phaeophyta (brown). Algae are often found growing in significant amounts along areas where there is little vegetation. The reason for this is that they are not dependent upon organic matter for nutrition. Algae increase oxygen and nitrogen* yields of the soil by photosynthesis and nitrogen-fixation respectively converts atmospheric CO_2 to cell substance and increases total organic carbon in soil. Nitrogen-fixation converts atmospheric nitrogen to organic nitrogen.

PURPOSE: To demonstrate the occurrence of algae in soil and to estimate the concentration in a soil quadrat.

MATERIALS: $NaNO_3$ - 10 g. ; $CaCl_2$ - 1.0 g. ; $MgSO_4$: $7H_2O$ - 3.0 g. ; KH_2PO_4 - 7.0 g; NaCl - 1.0 g. ; 15 test tubes, distilled or demineralized water.

PROCEDURE:

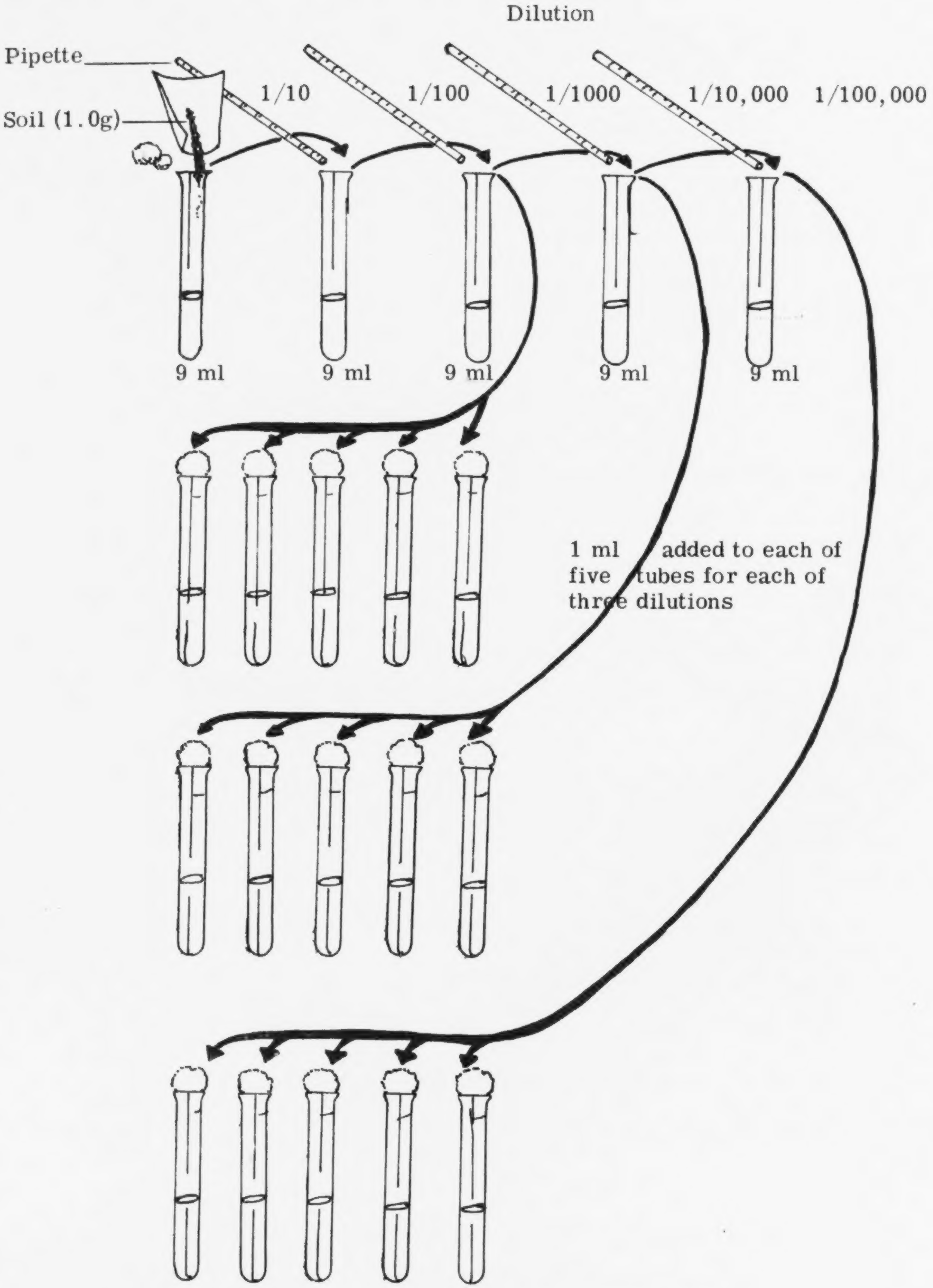
Part I - Stock Solution (prepared by instructor)

1. Mix the chemical compounds Na NO_3, $CaCl_2$, $MgSO_4$, $7H_2O$. KH_2PO_4, NaCl in the above proportions to 400 ml. of distilled water.
2. Add solution in procedure 1 to 940 ml. of distilled water.
3. Supplement with one drop of a 1.0 percent $FeCl_2$ solution.

Part II - Student Activity

1. Place 9 ml. of stock solution into 5 test tubes.
2. Add one gram of soil to the first solution.
3. Dilute the first solution as illustrated (See Fig. 1).
4. From the five original diluted solutions use the last three dilutions to prepare a series of three sets of five test tubes each.

*N - fixation is carried out by blue-green algae.


Dilution
Pipette
Soil (1.0g)
1/10
1/100
1/1000
1/10,000
1/100,000
9 ml
9 ml
9 ml
9 ml
9 ml

1 ml added to each of five tubes for each of three dilutions

Flow sheet for determination of abundance of algae in soil.

5. Incubate in diffused light for 30 days.

6. Observe results.

Part III - Algae Population

1. Using the solution from Part II, shake thoroughly and fill eye dropper. Drop one drop of solution from the dropper before placing the next drop on the slide.

2. Place 1 drop on a clean slide, cover with a cover slip. Count individual algae/algae colonies.

3. The method for the count is random; follow the diagram. Count algae/algae colonies in the areas marked 1 thru 5.

Colony Count number divided by five and multiplied by 39 and that answer multiplied by 1000 will give you the amount of algae per liter.

OPTIONAL: Count the green algae, blue-green algae, and diatoms separately and calculate the percent of each kind.

Make counts on the culture every day until they no longer show an increase in number. Graph this data with cell/liter on the vertical axis and time in days on the horizontal axis.

INVESTIGATION: COUNTING ALGAE POPULATIONS

1. Collect and Millipore filter a sample of test water as in Patch testing. If water contains an abundance of algae, it will be necessary to use a smaller sample, size, such as 10-50 ml.
2. When filtration is complete, remove test filter and allow it to dry for 30 minutes. Cut filter into four quadrants.
3. Place 5 ml of microscope immersion oil in a 47 mm Petri dish. Float the test filter on top of this oil. If filter has been completely dried, it will become transparent.
4. Draw filter across the edge of the Petri dish to remove excess oil; and center filter on a 2 "x3" microscope slide.
5. Scan the surface with low magnification. Count the number of algae in each of ten randomly selected fields in the filter.
6. Calculate the number of algae in the original sample using the following formulas:

   a. $\frac{1380\ (\text{mm}^2)}{\text{area of filed}\ (\text{mm}^2)}$ x number of fields counted = factor.

   b. Total number of algae counted x factor = number algae in sample. $1380\ \text{mm}^2$ = total filtration area.

   c. Number of algae in sample = number of algae per ml vol of sample (ml).

PROCEDURE: PATCH TESTING

1. After the sterifil holder and filter support has been sterilized, load the apparatus with a millipore type HA filter.

2. Add 250 ml of test water (keep volume of test sample constant from test to test).

3. Using the vacum pump, draw sample through filter.

4. Once filtration is complete, remove filter containing the trapped algal cells. Allow time to dry. Filter may then be mounted on chart along with other sample filters.

NUTRITION WEB

INVESTIGATION: SOLITARY OBSERVATIONS OF ECOLOGICAL INTERACTIONS

OBJECTIVE: To increase powers of observation and to become more aware of the continuous motion of living interactions in a community.

BACKGROUND: Most field trips to the school study site are characterized by lots of noise and confusion. Because of the constant interaction of teacher and student, many of the continuous interactions that occur in a natural community are missed. If the student were to sit quietly for 25 minutes he might be amazed at the living ecological interactions he may observe. The purpose of this exercise is to give the student this opportunity.

MATERIALS:
Pen
Paper
School site

PROCEDURE: The major requirement for this exercise is that the students *isolate* themselves from other *Homo sapiens*. When they have accomplished this, the instructions are simple. The students should be told something similar to the following:

1. Sit quietly for 30 minutes or until the teacher says time is over.
2. Listen for sounds. Record *all* sounds that are heard. Attempt an explanation of the source.
3. Record all smells that are encourtered. Attempt an explanation of the source.
4. Record all that is seen. When sitting quietly, many things that constantly occur may be observed for the first time. Birds may

even light on shrubs nearby. The behavior of an ant or a pill bug may become noticeable. Have the students try to determine what the animals are doing.

An interesting variation of this exercise is to have the students go into the school study site and follow an insect or other animal for 20 minutes. Have them record all the animal does during the time period, and attempt an explanation of the observed behavior. This can easily be accomplished by having each student find an ant hill at which solitary observation should be made for a designated period.

RESULTS AND DISCUSSION: After the trip is over, have the students write a paper indicating all the observations they have made. Have them attempt an explanation of each if they haven't already done so. As a summary paragraph have them evaluate the exercise in relation to making solitary observation as opposed to group exercise.

INVESTIGATION: FOOD WEB STUDY

OBJECT: To show the effect of eliminating so called "harmful" predators.

BACKGROUND: "Wanted . . . . Dead or Alive." Such signs are still to be found throughout the United States. However, they are aimed at certain animals that some men believe should be eliminated. These include such animals as wolves, coyotes, hawks, and mountain lions. Because these predators sometimes feed upon man's livestock, they have a "price on their head" in many states.

Once ocuppied by buffalo and sage hens, this region now supports cattle and chickens. The coyote, and some other carnivores, have shifted their feeding habits to include the new organisms. Farmers and ranchers claim that millions of dollars worth of livestock are destroyed annually by coyotes.

Supporters of the coyotes don't question the fact that they kill livestock. However, they suggest that the coyote does more good than bad by eating large numbers of rodents.

PROCEDURE: Study the data given and answer the following questions:

DATA:

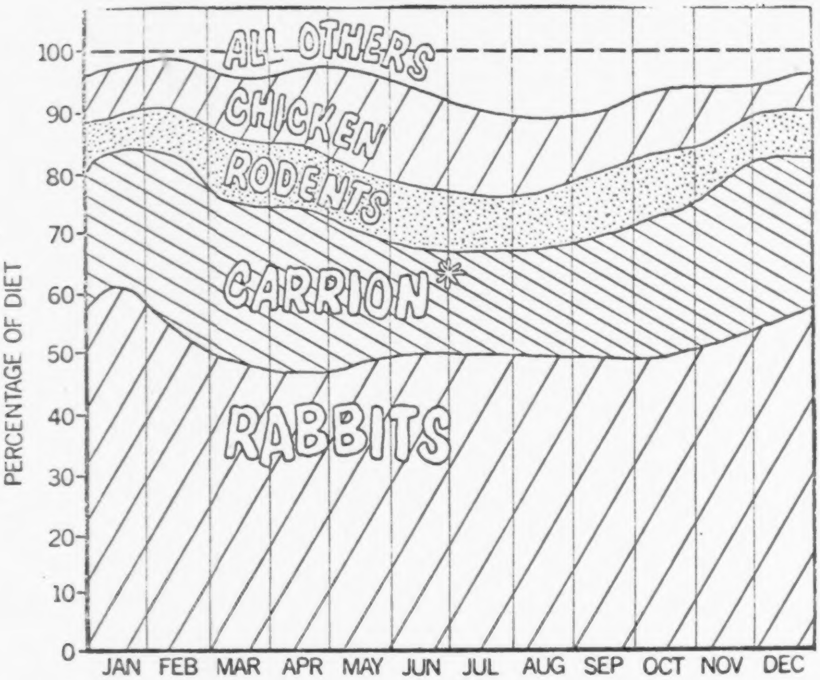


Figure 1

*Dead, not freshly killed meat.

Obviously, knowing something about the actual diet of the coyote would be helpful. Some of this information, for a midwestern state, is given in Figure 1.

Some estimates of the comparative amount of plant material eaten by rabbits, cows, and sheep are shown in Figure 2.

	NUMBER OF RABBITS THAT EAT AS MUCH AS	
	One Cow	One Sheep
Jackrabbits	75	15
Cottontails	250	50

Figure 2

According to Figure 1, coyotes also consume other animals that eat plant material--rodents.

Rodents the size of a meadow mouse eat about their own weight in grass daily. This amounts to about 11 kg of grass annually, or about one-half the daily grass intake of a cow.

QUESTIONS:

1. The main part of the coyote's diet consists of what organism?
2. What time during the year would the coyote's food habits interfere most with man's interests?
3. How do you account for the month-to-month variation in the coyote's diet?
4. What average monthly percentages of the coyote's yearly diet is made up of rodents?

How many rabbits does a coyote eat in a year? A few additional items of information will help you answer this question.

(a) An average of 53 per cent of the coyote's yearly diet is rabbits (mostly cottontails, a few jackrabbits).

(b) Coyotes require about two-thirds kilogram of meat daily, or about 250 kilograms a year.

(c) Cottontail rabbits weigh about one kilogram; jackrabbits weigh about 3. 3 kg.

(d) When rabbits are common, coyotes may not eat all of every one they kill. When rabbits are scarce, however, coyotes usually consume all of each catch.

5. How many rabbits do you estimate are eaten by each coyote anually? Cottontails? Jackrabbits?

Suppose we are considering a region that has a population of 25,000 coyotes.

6. How many rabbits would 25,000 coyotes eat annually? Cottontails? Jackrabbits?

That's a lot of rabbits. What does it mean in terms of plant material not eaten and, therefore, available for sheep or cows? Use the following formula to find out.

$$\frac{\text{Number of rabbits consumed by coyotes annually}}{\text{Number of rabbits that consume the food of one cow or sheep}} = ?$$

7. How many sheep or cows are not deprived of food by rabbits in the region each year? Sheep? Cows?

Let's not forget the mice and other rodents. Assume that each coyote eats 1,000 rodents each year.

8. How many rodents are eaten by the 25,000 coyotes annually?
9. How much grass is eaten by these rodents annually?
10. How much plant material does a cow eat? Daily? Annually?

If you had trouble answering the last two questions, you should review some of the earlier information in this excursion.

Use the following formula to answer question 11:

$$\frac{\text{Amount of grass eaten by rodents annually}}{\text{Amount of grass eaten by a cow annually}} = ?$$

11. How many additional cows can be fed annually by grass because coyotes consumed rodents?

12. How many cows can be fed annually because coyotes consume rabbits and rodents?

The large size of your answer to question 12 may have surprised you. Coyotes do a good job in keeping down the population of rabbits and rodents. Without this assistance from coyotes, a rancher might have to buy or lease much more land to raise the same number of cattle. Suppose it costs about $25 per cow each year to provide grazing.

13. What is the annual economic value to cattle ranchers of coyotes in the region being studied?
14. Do ranchers benefit more from the coyote's diet of rabbits or of rodents?

CONCLUSION:

Of course, this is only part of the story. Coyotes do eat chickens and occasionally young cows, sheep, and other domestic animals. How much are they worth? Is that economic loss offset by the other economic advantages?

Write, in your separate notebook, your conclusions concerning the pros and cons of coyote control. Should bounties be paid for dead coyotes or should it be made illegal to kill coyotes? Is there a good compromise between the two positions?

INVESTIGATION: FOOD WEBS IN FLORIDA COMMUNITIES

OBJECTIVE: To observe the animals of a living community and to arrange them in their probable trophic relationships.

BACKGROUND: While the concepts of food chain and food webs are easily understood from textbook reading and teacher lecturing, most students fail to see these living relationships in their surrounding environments. Cottontails, gopher tortoises, loggerheads, vultures, sea gulls, pelicans and rattlesnakes are common sights. This exercise is designed to follow classroom study of food web relationships or it may be used as the primary vehicle for understanding the concept of food webs.

MATERIALS: Animal and plant list
Cards
Paper arrows

PROCEDURE: This exercise can more easily be done after several field trips have been made to the school study site. At least one of the objectives of the trips should be to keep a list of the major (in numbers) plants and all the animal or animal signs. This list will be the primary source for this exercise (or alternatively one simply could elicite from the students animals they know are in the area. The former is preferred.) If the class has already completed a plant and animal survey of the school study site, the list then is no major task. Prepare for the students 4-6 sets of cards with each card bearing the name of a plant or animal observed (directly or indirectly) at the site. Also prepare small arrows to be used with the cards. If the classroom is equipped with tables, place the tables so that the students can work in groups of 4-6. Have the students arrange the cards into a food web for the community under study. Use the arrows to show that one organism is being eaten by another. It might be a good idea to have reference material available so that arguments can be settled by the students themselves. (The Ecology library comes in handy.)

RESULTS: Once the group has established their food web, have them defend it to the scrutiny of their peers. This can be done by having the groups pair off and compare their "masterpieces," allowing any group to make any changes necessary. The final product can be compiled by the class as a whole. The information should be recorded by the students for later additions and alterations.

DISCUSSION: (For the students)

1. Make a list of all producers, 1st order consumers, 2nd order consumers, 3rd order consumers and 4th , if any. Is the list easy to compile or is it difficult to designate a single slot for each animal? Why?

2. From your memory, which organisms seem to be the most numerous in your study site? To what trophic level do they belong? Are the most numerous organisms at the beginning or end of the food chain in which they are involved? Why?

3. Which animals on your list are there only on the basis of evidence of their presence and not by direct observation? Are these animals rare, or is there some other reason they are not easily observed? What reasons can you think of?

CONCLUSION: (for the students)

Make a general statement of the food relationships between the organisms found in your school study site.

INVESTIGATION: PHOTOSYNTHETIC PRODUCTS

OBJECTIVE : To determine the presence of starch and sugar in the leaves of plants.

BACKGROUND: During the daylight hours starch accumulates in the leaves of green plants; by evening a relatively high concentration is present. During the night this starch disappears from the leaf. The starch is reconverted to glucose and carried away in the veins to all parts of the plant. The starch is insoluble in water, thus the need for its reconversion to glucose for transportation. Thus the starch is transported as a sugar solution to various parts of the plant to provide food for growth and for storage. Food must be carried to all the underground parts, to the cells deep in the interior of the stem, and to the flower parts. There are special tubular cells called sieve tubes in the veins that conduct not only sugar but other soluble organic foods, like amino acids and fatty acids. Thus, in the veins of the leaf there is a two-way conveyer belt, an upward movement of water and a downward movement of factory products in the sieve tubes.

Starch is formed in the leaves only when the glucose resulting from photosynthesis reaches a certain concentration. Early in the morning, therefore, leaves contain little or no starch, the amount reaching an appreciable level by afternoon. Starch tests on leaves are best carried out in the late afternoon, or evening.

PART A

The addition of iodine to a substance containing starch causes a definite color change. No other substance exhibits this color change in the presence of iodine. To enable you to identify this characteristic color change add a few drops of iodine to a raw or cooked potato and observe the color change. Now you are ready to test leaves for the presence of starch.

1a. One cannot test a leaf for starch by dropping iodine on a freshly cut leaf. There are complications. First the iodine cannot penetrate the living cells where the starch grains lie and secondly, the chlorophyll present in these cells interferes with the color change. Thus the cells must be killed and the chlorophyll removed.

Select two leaves, one from each of the lists A and B.

A

Tomato
Bean
Geranium
Any deciduous tree
  or shrub, e. g.
  maple

B

Grass
Corn
Iris
Onion

Remove leaves from the plant after they have had at least six hours of sunlight. Dip them in boiling water, which kills the cells, enabling the iodine to enter them. Now extract the chlorophyll by immersing leaves in hot rubbing alcohol. When leaves are a creamish color, remove them from the alcohol, wash them and place them on a white surface and add iodine. Rinse off excess iodine and compare the color of the two leaves. Which contains starch?

2b Repeat preceding experiment using leaves picked early in the morning and late in the afternoon. Compare starch content of early morning leaves with that found in leaves collected late in the afternoon.

PART B (test for presence of sugar)

If you are acquainted with diabetes you probably know that there is a chemical preparation which can be used to identify glucose sugar in urine. This preparation (Clinitest) can also be used to identify simple sugars, for example, glucose and fruit sugar (fructose) in leaves.

2a. Clinitest and Common Substances

Drop one Clinitest tablet into each of the following, contained in a glass vial.

1. One-fourth teaspoon of honey dissolved in one tablespoon of water.
2. One-fourth teaspoon of ordinary sugar dissolved in one tablespoon of water.
3. One-fourth teaspoon of cornstarch mixed with one tablespoon of water.
4. One tablespoon of raw egg white (pure protein).
5. One tablespoon of tap water.

Note the color change in each case (refer to the color chart included with the Clinitest preparation); 5 acts as a control.

EXPLANATION:

Only honey, a mixture of glucose and fructose, affects the blue color of the watery Clinitest preparation, turning it orange. If leaves give this same result, we would assume that they also contain glucose and/or fructose.

2b. Using the Clinitest Method on Leaves

Collect some leaf samples from plants listed under category A and B.

Chop them finely and ground them with a mortar and pestle. Add a little water; strain about 3 ml. into a vial or test tube. Add a Clinitest tablet and observe. Do both sets of leaves contain simple sugars?

EXPLANATION:

The explanation of results lies in the fact that there are two types of leaves, those which in the presence of light convert simple sugars into starch (when the concentration of these reaches a certain level), and those which never form starch, although they may convert simple sugars into complex sugars like sucrose.

Starch Leaves	Sugar Leaves
Deciduous trees and shrubs	All grasses, including: sugar cane, cereals (e. g. rice, corn, wheat)
Tomato, carrot, bean, pea, lettuce, potato	Bulbous plants (e. g. onion, garlic, lily, iris)

It is interesting to note that the members of each group have other features in common, for instance, a similar flower structure, indicating phylogentic relationship.

PART C Starch Production in Variegated Leaves

The variegated geranium leaf, which shows a margin of white around a green center is a good subject. Any other variegated plant is suitable, as long as it is not included in the list of sugar leaves.

Remove leaves to be tested in the late afternoon and make a careful diagram to show the distribution of the green and white areas. Using the procedure outlined in Investigation 1a, test each leaf for starch. Compare the distribution of starch with the distribution of the green and white areas. What are your results?

INVESTIGATION: GENERAL SOIL BACTERIA

OBJECTIVE: To observe the presence and numbers of bacteria in Florida soils.

BACKGROUND: Bacteria play a very important role of decomposers in soil. Their presence may be over looked simply because they are microscopic. They may be observed qualitatively by plating bacteria from soil samples taken from the area of study.

Some of the most common bacteria of soils are the actinomycetes. They are unicellular organisms that produce hyph, thus causing some taxonomist to classify them as fungi. The most common species are of the genus Streptomyces (actinomycetes are the natural source of many of our antibiotics. These chemicals are produced naturally as an aid to competition for food). These streptomycetes have an earthy odor resembling freshly plowed earth which can be detected in the petri dish.

MATERIALS:
- Petri dishes
- Soil samples
- Test tubes of nutrient agar or soil extract agar
- Innoculating loops
- Test tube racks
- Pipettes (1 ml or 10 ml)

PROCEDURE: Carefully weigh out 1.0 gram of soil to be tested and transfer into 9.0 ml of agar at 45^{o} C. Shake the tube to disperse the soil. Using a sterile pipette, transfer 1.0 ml from the tube into a test tube #2 of 9.0 ml agar, that has been cooled until it can be held in the hand.

(Nutrient agar may be purchased from a supply house.

Soil extract agar may be prepared as follows:

Add 500 grams of soil to 1500 ml of tap water. Autoclave at 15 pounds for 30 minutes. Filter through cloth or paper until clear. Prepare the following.

Soil extract - 500 ml
Tap water - 500 ml
Mannitol - 5.0 gm
K_2HPO_4 - 1.0 gm
Asparagine - 0.1 gm
Agar - 15.0 gm

Sterilize in autoclave or pressure cooker at 15 pounds for 20 minutes.)

Tap the tube on the base of the palm of your hand to disperse the soil. Quickly before the agar solidifies, transfer 1.0 ml into another tube that has been cooled. Pour tube #1 into a sterile petri dish while your lab partner transfers 1.0 ml of tube #2 into tube #3. Pour tube #2 into a sterile petri dish and repeat the procedure until four dishes have been poured. Label Dish #1-1/10, Dish #2-1/100, etc. See diagram of procedure.

RESULTS: Allow the agar to solidify and incubate the plates at 28° C. for 7 days. Examine the plates noting the difference in the number of colonies in the different dilutions. Choose the one plate with 30 to 300 colonies. Make a total count distinguishing other bacteria from the actinomycetes. (See diagram of act.) Record other bacteria according to shape of colonies and pigmentation. Observe under the microscope to distinguish bacteria from actinomycetes. See Chart Page 4-19.

CALCULATIONS:

1. Total # of bacteria and actinomycetes per gram of soil

   Total count of bacteria x dilution and total count Actinomyces x dilution

2. Total per cent Bacteria

   Total count ÷ into total bacteria x 100

3. Total per cent of actinomycetes

   Total count divided into total actinomycetes x 100

CONCLUSION:

Make a general statement about the kinds and abundance of soil microbes in your sample.

DATA: After 7 days, observe the dishes for fungal growth. Make sketches of the growth, noting different shapes and colors of the colonies. Record the data in the following chart. You may want to use the microscope to enhance observation.

Dilution	No. of Different Kinds	Total No. of Fungi	No. per Gram
1/10			
1/100			
1/1000			
1/10,000			
1/100,000			

CHART OF RESULTS

TOTAL COUNT	OTHER BACTERIA Color & Shape			ACTINOMYCETES
	#1	#2	#3	
Plate # 1 1/10				
Plate # 2 1/100				
Plate # 3 1/1000				
Plate # 4 1/10,000				

INVESTIGATION: A STUDY OF FROG PARASITES

BACKGROUND: Green plants produce food. Herbivores consume plants and carnivores consume other animals. In another kind of nutritional relationship called symbiosis, individuals live in direct association with one another. Parasitism is a common form of symbiosis. In parasitism the parasite benefits while the host is harmed. Every free-living organism appears to have its parasites. Many of them have more than one kind of parasite. The common frog can be used to study parasites because it often has them on its skin, between the skin and muscle, and in various tissues in its body.

MATERIALS:
- Live frogs
- Microscope slides
- Cover glasses
- Large-mouth jar or plastic bag for etherizing frogs
- Cotton
- Dissecting tray
- Scapel
- Scissors
- Forceps
- Dissecting needles
- Straight pins
- Petri dishes
- Ethyl ether
- 0.7% saline solution
- Methylene blue

PROCEDURE: Examine the frog's skin to look for ticks. Study the structure of the ticks under the microscope, noting the type of mouthparts and presence or absence of appendages. Make a sketch of specimens.

Etherize frog by putting it into a large-mouth jar or plastic bag containing cotton soaked in ethyl ether. When the frog is limp, remove it from the container and place it ventral side up in a dissecting tray. Pin it to the tray by stretching its legs and inserting a pin in the front and hind feet.

In examining all of the parasites you find in the frog, prepare a slide by putting a few drops of saline solution on the slide, then add the tissue containing the parasites. Put a few drops of methylene blue on the tissue to stain the parasites for better study. Cover with a cover glass and examine under low or high power, depending on the size of the parasite. Make a drawing of all the parasites you find. Be sure to note whether the parasite is attached by means of a special organ or free. If free, note the means of locomotion. Determine the presence or absence of a mouth, digestive tube, and anus. Look for reproductive organs and determine whether the sexes are separate or the animal is an hermaphrodite.

Cut off one of the fingers and collect a drop of blood on a microscope slide. Cover with a cover glass and examine under high power of the microscope. Search among the red blood cells for small worm-shaped organisms, the embryonic nematodes. If these are present in the blood, you can expect to find some of the adults between the skin and muscle wall. Make a longitudinal cut along the trunk with your scissors. Look for the adults between the skin and muscle wall, in the abdominal and axillary regions, or in the thoracic and abdominal cavities.

Remove the entire contents of the thoracic and abdominal cavities. Separate the organs and put them in individual petri dishes containing a small amount of saline solution. Pull the lung and liver tissue apart with your dissecting needles. Examine with the naked eye for the presence of round worms and flat worms. Then examine some of the tissue under the microscope for other parasites.

Cut open the stomach and intestine and examine the contents with the naked eye and with a microscope for the presence of parasites. Examine the contents with a microscope for the presence of parasites. Examine the contents of the urinary bladder and the bladder tissue with the naked eye and with a microscope for flat worms and round worms. Cut open the rectum and mount bits of the fecal wastes for microscopic examination.

If you wish to further your study of symbiosis, you may examine the intestine of the termite for cellulose-digesting flagellates that live there or examine the seminal vesicles of the earthworm for protozoans. You might also collect materials from ponds and look for green hydras or paramecia that have green alga living with them in a symbiotic relationship.

AQUATIC ECOLOGY

ITEMS TO BE CONSIDERED AT THE SITE

Aquatic site (home study or school site-individual)

1. Temperature, humidity
2. Phosphates
3. Nitrates, nitrites
4. Detergents
5. Biochemical oxygen demand (BOD)
6. Coliform bacteria
7. Sewage effluent
8. Photographic documentation of area used
9. pH
10. Generalized qualitative survey of the flora and fauna
11. Rainfall
12. Shade, cloud cover, exposure
13. Bottom composition
14. Plankton
15. Benthic invertegrates
16. Succession (growth and development)

AQUATIC ECOLOGY - DATA SHEET

Physical Data

Date of sample													
Time (AM, PM)													
Depth (inches)													
Water temp. °C													
Air temp °C													
Wind direction													
Precipitation (amt. during past 6 days)													
Cloud cover (48 hours) or shade													

Bacteriological Analysis

Date of sample														
Coliform 1/10														
Coliform 1/100														
Coliform 1/1000														
Coliform 1/10000														

Chemical Analysis

Date of sample															
Nitrates (ppm)															
Phosphates (ppm)															
Dissolved 0_2 (ppm)															
BOD (ppm)															
Detergents (ppm)															

Algae and Protozoa

Cells /ml)

Date of sample															
Green algae															
Diatoms															
Dinoflagellates															
Blue-grn. algae															
Protozoa															

INVESTIGATION: WATER SAMPLER CONSTRUCTION

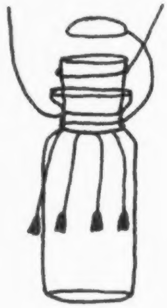
PURPOSE: Construction of a water sampler.

MATERIALS:

250 ml to 300 ml reagent bottle with a ground glass stopper
Centimetercalibrated cord
Six inch thin wire
Some 1 oz. pyramid sinkers
Green and red yarn or string

PROCEDURE:

1. Bend the six inch wire around the neck of the reagent bottle and bend the remaining wire into a circle a half inch above the stopper as illustrated.



2. Calibrate cord into units of decimeters and half decimeters. Do this by tying green yarn or string around the 10 centimeter marks and red yarn or string around the five centimeter marks. Leave a little cord uncalibrated in order that it can be tied around the neck of the bottle. This string will be used to lower the bottle and measure water depth.

3. Around the neck of the bottle tie a cord with pyramid sinkers hanging off and distribute the sinkers around the bottle. The sinkers will

give the empty bottle weight enough to sink. Add more weight as needed. Secure sinkers to the bottle by means of some waterproof tape, such as fiberglass tape.

4. Tie a cord to the stopper of the bottle. Cut this cord as long as the calibrated cord. This cord attached to the stopper will raise the stopper at the depth from which the sample is to be taken. The sample will then be ready for phosphate test and any other tests used in water analysis.

INVESTIGATION: STREAM OR CANAL FLOW

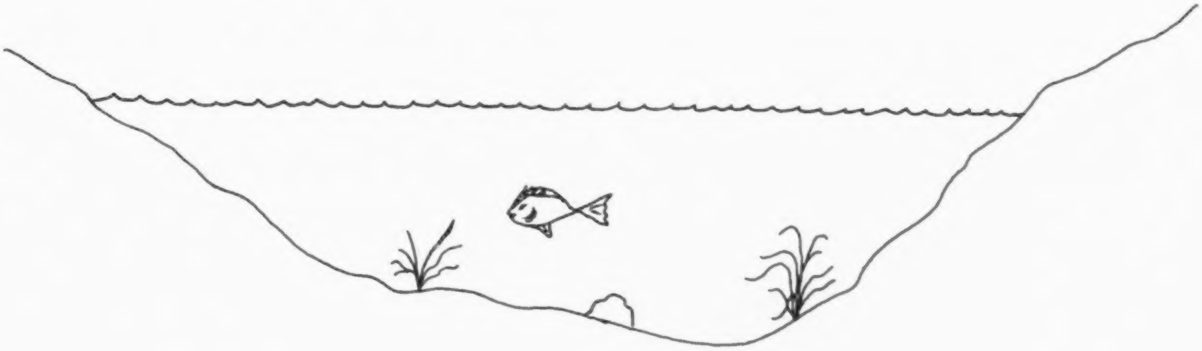
BACKGROUND: Stream flow or total volume passing any given point on a stream can be calculated using the formula for discharge. In order to be valid, discharge data must be collected over a period of time especially if it is to be linked to effluent or sewage data. The reason being no stream flows consistently in volume. This leads to variations in sewage discharge data and effluent data. Long-term observations present a truer picture of the relationship between stream contents and flow.

There are streams whose flow consists of 90% sewage effluent during certain times of the year and only 5% sewage effluent at other times. Because of this variation, stream flow must be determined and recorded for all studies carried out over an extended period of time.

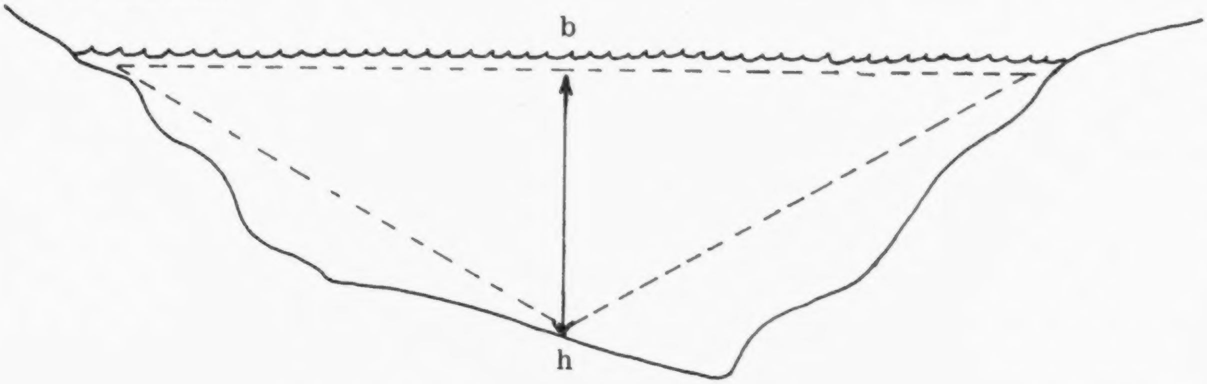
Stream flow is defined as the volume of water passing a point during a given period of time. The U. S. Geological Survey maintains over 6, 000 stations in the U. S. where stream flow is gauged. The equation used to calculate stream flow is $Q = AV$ where Q = discharge in cubic feet per second, A = channel cross section at the sampling point, and V is mean velocity at the cross section site.

The velocity is calculated by inserting the velocity measuring instrument just under the water surface in approximately the middle of the stream or any spot which is not affected by obstructions (rocks, logs, or docks) and is also away from the stream banks. (The instrument must be calibrated in feet per second for use in calculating stream flow.)

Area is calculated by finding depth at the middle of the stream and the distance across. The formula for the area of a triangle is then used to calculate the approximate area of the cross-section:



Stream x-section: b (base) = width of the stream and h (height) = depth of the stream.



Area = 1/2 bh

This figure is inserted as area of cross section (A). Now Q (stream flow) can be computed using the already cited formula Q = AV.

Using these parameters the amount of a particular pollutant contributed by this stream or river to another body of water can be computed.

INVESTIGATION: SHORE-LINE SURVEYS OF PONDS

In the course of ecological investigations, it may become necessary to determine precise points along the shore of a small lake or pond. Generally, topological maps are not made on a small enough scale to encourage their use for bodies of water in the order of less than one acre to those of fifteen or twenty acres. It then behooves the investigator to prepare his own map on a scale as is accurately possible. This exercise is for the purpose of enabling the student to become proficient in the use of the compass (or transit), alidade, stadia rods, plane table, and the mechanics of constructing a map to be used in his investigation.

"A series of connected straight lines whose lengths and angles have been determined by appropriate methods and instruments is known as a traverse." This particular method is especially useful where all points of the shoreline are not readily accessible from the shore.

The equipment for this exercise need not be so sophisticated that the student loses interest before beginning his investigation. A small compass, a 100-ft. steel tape, sufficient wooden stakes to be driven as reference points, thumb tacks, 2 or 3 range poles and a form for records.

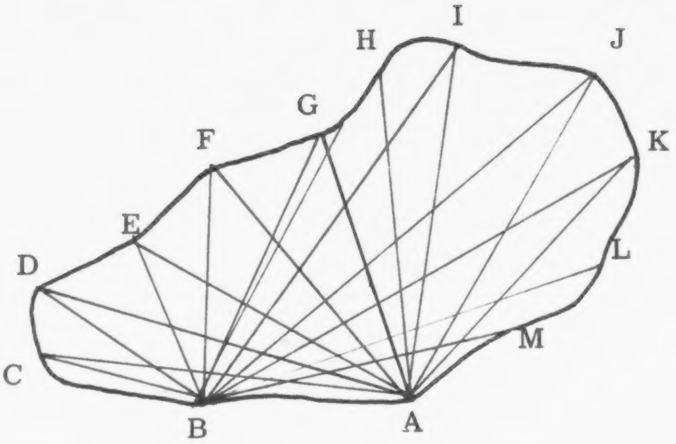


Figure 1 - Diagram illustrating location of positions on the shore line of a pond or small lake by method of simultaneous measurement of two angles and intersection of lines. (AB) base line of known bearing and length. (C-M) shore line stations. Radiating straight lines indicate lines of sight from one transit to intersection with corresponding lines of sight from other transit.

PROCEDURE: (Minimum personnel consists of two people: one to remain at the base line for reading and the other to establish targets at the appropriate points.) A preliminary investigation of the site is in order so that the plan of survey may be determined; shore line details must be examined in order to establish the most favorable position for base line and transit positions. Transit stations should be chosen in such a manner that all pairs of lines prolonged from transits will form angles at least greater than 30 and less than 120 . After establishing base line (AB), rod man proceeds to point C and places rod upon stake and attaches a small white flag to base stake; flag must be kept on stake until released by signal from base transitman. Rod man then proceeds to each of following stakes until sightings have been taken on all stakes from point A; the procedure is repeated from point B. All sightings are recorded on the appropriate form. Rod man should keep log of each station including brief description of same. Having taken all readings, information may now be translated into map on graph paper of suitable scale.

FIELD RECORD

Survey of Shore Line of ____________________ by ____________________

Rod man ______________________________ Date ____________________

DATA

Stake	Angle from Transit A	Angle from Transit B	Magnetic Bearing		Descriptive Remarks
			Transit A	Transit B	

INVESTIGATION: AQUATIC MICROCOSMS

BACKGROUND: Aquatic communities undergo change in composition with time similar to the successional changes seen in terrestrial communities. The temporal changes seen in aquatic environments and the rates of change are often closely related to the availability of particular chemical nutrients. In a pioneering aquatic community the first organisms to appear can be either producers (autotrophs) or consumers (heterotrophs) depending upon whether the nutrient supply is primarily inorganic or organic respectively. Inorganic nutrients encourage the growth of autotrophs (certain bacteria and algae) while organic nutrients encourage heterotrophs (fungi, saprophytic bacteria, and protozoa). Microcosms (small isolated ecosystems) change in species composition with time in a manner seen in larger ecosystems, but are more convenient to study. Protozoa feed upon bacteria and algae while rotifers and predaceous aquatic insects feed upon both of these species resulting in an increase in species diversity and population stabalization. An excess of either organic or inorganic nutrients can inhibit or reverse the development of the community. Clear under-feed waters are said to be oligotrophic and represent early states in aquatic succession as opposed to the over-feed eutrophic condition which can result in pollution. It is possible to simulate and study these conditions under the controlled conditions in the laboratory and to investigate and/or demonstrate the principles of aquatic ecosystem development or death by additions of excessive nutrients or toxic substances.

PURPOSE: To learn the principles of ecological succession and its relationship to nutrient or toxic conditions in the environment.

PROCEDURE: Four or more beakers or jars of equal size should be half filled with water (see additions: Nutrients).

Additions of nutrients, detergents, herbicides, or insecticides can be made to the beakers if a single source of water has been used. The amounts of the additions to be added to the beakers should first be determined by testing a series of dilutions for their toxicity. Nutrient additions are made in the form of commercial liquid fertilizers in amounts specified on the label. Additions of single nutrients, e.g., phosphates, nitrates, from the chemical stock room can be made either singly or in combination.

Additions:

I. Nutrients

A suitable combination of nutrients can be provided by additions of liquid fertilizers. The amounts of these additions can be those prescribed on the label or variations above or below these recommended amounts. The basic nutrients can also be enriched (entrophicated) with solutions of phosphates, nitrates or organic compounds (sugar, peptone, urea, etc.).

In fact, a synthetic waste water stock can be made as follows:

1 liter H_2O
16 g glucose (dextrose)
16.5 g peptone*
2.5 g urea
10 g $NaHCO_3$

Dilute 10 ml of the stock solution with 1.0 liters of tap water.

II. Detergents, Insecticides, Herbicides and PCB**

The toxic nature of these compounds can be demonstrated by their addition to the microcosms in a series of dilutions. That is, one ml to the toxin to nine milliliters of water (1 in 10), then one milliliter of this solution is added to nine milliliters of water (1 in 100) and so forth until the toxic effect is diluted away. Once nontoxic levels are determined various brands of the substance can be compared by controlled experimental procedures.

The biodegradability of these compounds can be tested by making these additions to the synthetic waste water preparation. After activation*** with air for two days samples can be tested in microcosms for toxic effects or chemical alteration.

III. Organisms

The water used in these experiments can be distilled or de-ionized water, lake water, river water, or canal water. If distilled or de-ionized water is used, a small measured quantity of natural water can be used as a source of planktonic and sessile organisms. A small sprig (3 1/2 in.) of the growing end of an Elondia or coontail moss plant can be added to each microcosm. These plants grow rapidly in unpolluted enriched environments. The sprigs should be carefully measured every other day and the data graphed (length vs. time). Animals, small fish or snails, are also good biological indicators of conditions in these little aquatic worlds.

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*Gives a final concentration of 3 mg/liter phosphorus

**PCB, polychlorinated biphenyls = derived from pasticizers and other industrial chemicals.

***See section 1-20 on sewage disposal.

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Mitchell, Dee, Eutrophication of Lake Water Microcosms: Phosphate Detergents, Science, 174:827 (1971).

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INVESTIGATION: FRESH WATER INVASION OF A SALT WATER ENVIRONMENT

INTRODUCTION: This investigation is intended as an exercise in which the student can put many of his lab-learned practices into field use. It is also intended to give the student an opportunity to increase his powers of observation by making close studies of two environments and then drawing conclusions from his observations. Also by comparing his observations with others, he will have an opportunity to see how complete his facility for observation is. It will also show him how he might improve them.

BACKGROUND: Much concern is sometimes expressed on the problem of salt water invasion of fresh water areas. Salt water invasion is seldom given a passing thought. Many results are common in both instances insofar as marine life is concerned. One difference is that Man is usually affected severely by fresh water invasion hence, little is heard about it. Conservation is, however, important and fresh water invasion is primarily the result of man's work therefore, the adaptability and survival of salt water organisms should be investigated.

PURPOSE: To study the effects of fresh water invasion of a salt water environment.

MATERIALS:

Materials for the determination of salinity
Materials for the determination of suspended solids in water
Recording notebook for observations
Water containers (50 ml minimun)
Thermometer

PROCEDURE:

PART I

1. Record that date and time of observations and also record the weather and tide conditions if applicable.

2. Make detailed observations of life forms. Note especially the size and condition of algae and fish.

3. Take water sample.

4. Record water temperature.

PART II (Back at Lab)

1. Determine the salinity of the salt water by use of the refractometer. If a refractometer is not available determine the salinity by titration.

2. Determine the weight of the suspended solids by filtering the water sample.

PART II (After invasion)

1. Repeat steps 1-4 in Part I and Part II.

2. Note any organisms which were also present before the invasion. (Size and condition of such.)

INTERPRETATION OF DATA

1. What were the differences in the algae and fish population and size between observations?

2. How could you explain the presence of any specific organisms in

each observation?

3. Did the fresh or salt water have a higher weight of suspended solids? Explain.

4. Compare the color, temperature, and amount of trash found in the water at each observation.

5. Were there any differences in the condition of the shore line between observations? If so, give a possible explanation.

6. Was there any evidence in the observations that might indicate any chemical pollution?

7. If necessary, how could fresh water invasion be stopped?

8. How might an invasion of fresh water hurt an area commercially?

INVESTIGATION: ORGANISMS OF THE BENTHOS

PURPOSE: To determine the amounts and kinds of benthic aquatic invertebrates.

MATERIALS:

- Dredge
- Mason jars
- Rakes
- Nets
- Soil sieves

PROCEDURE:

PART I: Qualitative (variety)

In some environments it is possible only to collect qualitative samples because the physical nature of the waterway may be such that quantitative sampling is not feasible. The qualitative search for benthos (bottom dwellers) should invlove the collection of organisms from rocks, plants, submerged twigs or debris, or leaves of overhanging trees that become submerged and waterlogged. It is often convenient to scrape and wash organisms from these materials into a bucket or tub partially filled with water and then to pass this water through a sieve to concentrate and retain the organisms. The collected sample may be aereated for identification later.

Qualitative sampling determines the variety of species occupying a reach of a waterway. Collections from such samplings indicate changes in the environment, but they generally do not accurately reflect the degree of change. Mayflies, for example, may be reduced in the stream because of adverse conditions from 100 to 1 per square foot, whereas sludgeworms may increase from 1 to 14,000 per square foot. Qualitative data would indicate the presence of both species, but might not necessarily delineate the change in predominance from mayflies to sludgeworms.

Two convenient limiting methods for qualitative sampling are: (1) Presetting a time limit on the collector's effort at each sampling point. A minimum of 30 minutes and a maximum of an hour is a convenient range. (2) Sampling in an area until new forms are encountered so infrequently that "the law of diminishing returns" dictates abandoning the sampling point.

A number of tools readily obtained by most anyone are valuable in qualitative sampling: (a) Pocket-knives are excellent tools to remove animals from crevices in rocks, to peel bark from decaying logs thus exposing animals, and to slip under animals to lift and transfer them to sample containers. (b) Mason jars in 1/2 pint sizes serve as the most economical sample containers and provide visibility of the specimens. (c) Common garden rakes are valuable to retrieve rocks, brush, logs and aquatic vegetation for inspection. (d) Fine-meshed dip-nets are good devices for sweeping animals from vegetation or out from under over-hanging ledges. (e) Buckets are handy to quickly receive rocks and debris, thus preventing escape of the swift running animals. (f) Sheet polyethyelene, 6 x 6 feet, can be spread on the stream bank and substrate materials placed upon it. As the materials begin to dry the animals will abandon their hiding places and can be seen readily as they migrate across the sheet seeking water. (g) U. S.

Standard Series No. 30 soil sieves can be used to scoop up fine sediments and sieve out its inhabitants. (h) Any other tools, such as forceps, scapels, shovels, and forks are ligitimate devices, and can prove their merit in individual situations.

PART II: Quantitative (total number)

Following these general observations, the investigator collects appropriate quantitative samples of the various kinds of organisms present in the aquatic area. He makes certain that, (1) the sampling area selected is representative of stream conditions, and (2) the sample is representative of and contains those forms predominant in the area and encountered during the qualitative search.

Samplers such as the Ekman dredge or Peterson dredge are excellent. If commercially made samplers are not available, a simple device using a weighted can on a rope may be used. In more shallow areas the can may be suspended from the bottom of a stick in a hinged fashion to make it more manageable. Collecting technique must be refined in order that samples of similar size may be taken.

Artificial substrates are placed in the water for 3 to 6 weeks and then carefully removed to prevent losing the organisms that have made them a temporary home. As nearly as possible the substrates should be placed at similar depths and in similar physical relationship to the stream at all stations. Usually they are placed about 1 foot beneath the surface and 1 foot off the stream bed.

The type of artificial substrate employed to collect organisms is not terribly important as long as the same type is used at all such sampling stations in a particular investigation. Any type will be somewhat selective by those organisms that are attracted to it. They do tend to favor drift organisms or those that become detached from their dwelling areas and float downstream with the current. When the same type of sampler is used at each station, data collected among the stations should be comparable.

NOTE: Fish, such as bream or bass, are good indicators of well oxygenated water; however, gar and *Gambusia* gulp surface air and therefore can withstand poorly oxygenated waters.

REFERENCES:

Gaufin, A. R., Tarzwell, C. M., *Aquatic Macro-Invertebrate Communities as Indicators of Organic Pollution in Lytle Creek*, Journal of Sewage & Industrial Wastes, Vol 28, pp. 906-924.

Morgan, *Field Book of Ponds and Streams*.

INVESTIGATION: MICROSCOPIC FORMS IN THE SAND

BACKGROUND: This exercise is designed to take 3 or 4 class periods. The exercise on agar digesters and luminescent bacteria can be run concurrently by extending the time to 5 days + 1 day.

It is advisable to have the required media prepared ahead of time in tubes containing 5 ml each, autoclaved and stored in the refrigerator until needed. If the media are stored in test tubes with approximately 5 ml per tube, the student can melt a tube of agar and pour the required petri dish, medicine bottle, or make an agar slant. In addition, this procedure saves time when each student or team of students does not have to prepare his own media.

Make a trip to the beach and remove an area of sand at the low tide line approximately 2 cm deep and 30 cm square. Place the sand in a bucket and return to the laboratory.

A deserted beach appears to be a relatively sterile area, devoid of life and offering little or no protection or nutrition. If we examine the beach more closely, we would be surprised at the abundance of life forms that exist.

Many of the life forms are microscopic bacteria, yeasts, and molds. A great many of these forms are attached to grains of sand or bits of shell. On further investigation we should realize that every time the tide changes some organisms are added and some are washed out of the beach sands.

PURPOSE: To ascertain the presence of bacteria, molds, and yeasts in beach sand.

MATERIALS:

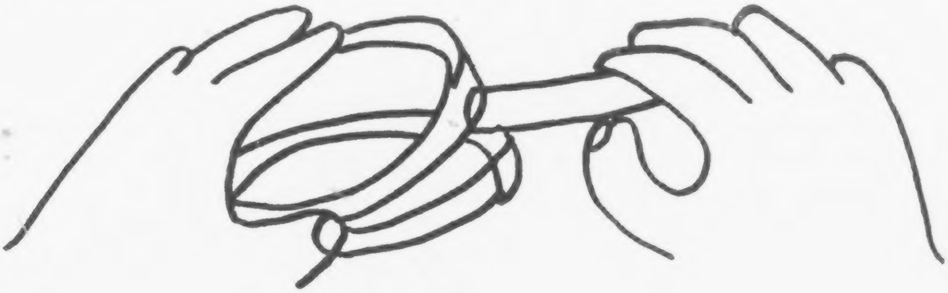
- 12 petri dishes or 3 oz. medicine bottles
- Bensen burner or alcohol burner
- 12 slides and cover slips
- Compound microscope
- Crystal violet stain 2%
- Sterile sea water
- Inoculation loop or sterile "Q-tips"
- Grease pencil
- Marine bacteria agar medium
  - 1 liter sterile sea water
  - 10 grams peptone
  - 15 grams agar
- Marine yeast agar medium
  - 1 liter sterile sea water
  - 20 grams agar
  - 23 grams dextrose
  - 1 gram Sol-U-Pro or equivalent protein
  - 1 gram yeast extract
  - 100 ml chloromycetin or terramycin
- Marine mold agar medium
  - 1 liter sterile sea water
  - 17 grams agar
  - 1 gram yeast extract
  - 10 grams dextrose
  - 100 ml chloromycetin, terramycin, or pencillin

To prepare the media, the water should be brought to a simmer, then the agar and other ingredients added and stirred until they dissolve.

PROCEDURE:

1. Prepare a minimum of 3 petri dishes or medicine bottles of each type of medium by the method illustrated. After melting a test tube of the desired medium, the tube should be held in the right hand with the petri dish lid opened by the left hand. The lid should be opened only enough to allow

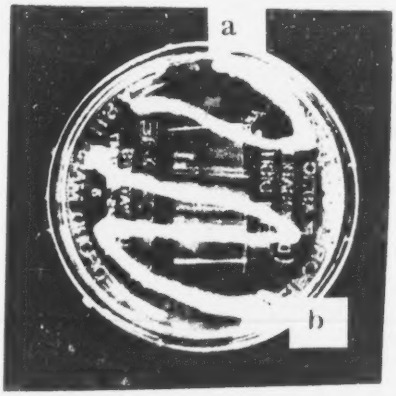
the mouth of the tube to be inserted and its contents emptied into the petri dish and then closed as rapidly as possible to prevent contamination. The entire process should be carried out in a smooth flowing motion rather than a hurried jerking motion. Allow the medium to harden.



2. Add 1/3 tube of the sand sample to 1/3 tube of sterile sea water; stopper and agitate thoroughly.

3. Permit the sand to settle in the tube, then decant 1 ml of the liquid into a sterile tube for use.

4. Using the inoculation loop or "Q-tip", streak two of the three petri dishes from each type of agar as illustrated (one dish from each group is to be kept as a control). Begin at position "a" with the inoculating loop and lightly draw the loop across the agar surface in the illustrated pattern, being careful not to break the surface of the agar. On finishing at position "b" the plate should be closed immediately and the inoculation loop flamed.



5. The inoculation loop should be passed through a flame before and after each use. The mouth of a test tube is to be flamed before and after

each opening of the tube and the tube held at a downward angle, when open to prevent contamination from air currents.

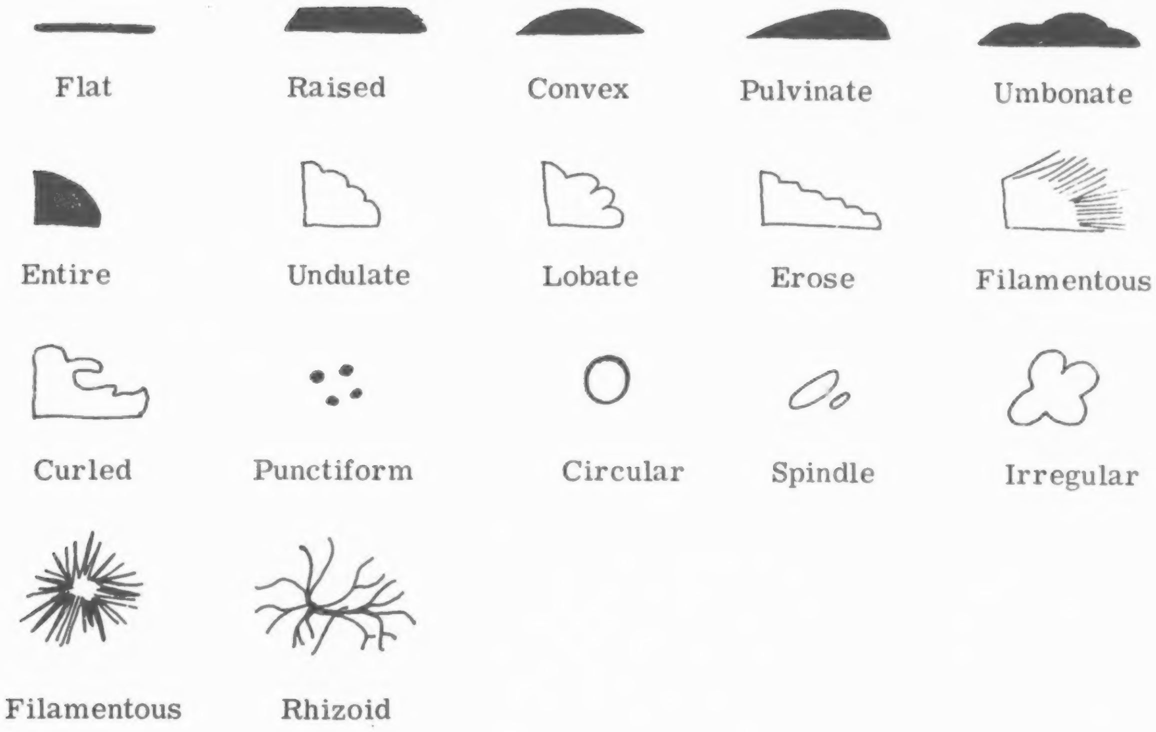
6. The dishes of inoculated agar plates are to be kept in a warm, humid atmosphere for the next 4 days, preferably in an incubator with a dish of water to provide humidity.

7. The following observations are to be made each day and recorded in the log: (a) number of colonies present, (b) color, shape, and size of each colony, and (c) growth pattern for each colony. From each type of growth on each plate, remove a sample of the colony with the inoculation loop and mix with a drop of sterile sea water on a clean slide. Cover with a cover glass and observe under both low and high power of the microscope.

8. After the initial observations, each slide is to be stained with 2% crystal violet by the following method: (a) remove the cover slip; (b) pass the slide gently through a flame 3 or 4 times or until dry with the set side up; (c) when the slide cools to room temperature, cover the slide with 2 or 3 drops of the stain; (d) allow the stain to remain on the slide for 30 seconds, then pour off the excess; (e) the slide should now be rinsed in a beaker of clean water by immersing gently; (f) the water left on the slide is removed by blotting gently with a paper towel.

9. The slides are now ready for observation under the microscope.

10. List and sketch the organisms observed at this time. Identify each according to the media it was grown on.

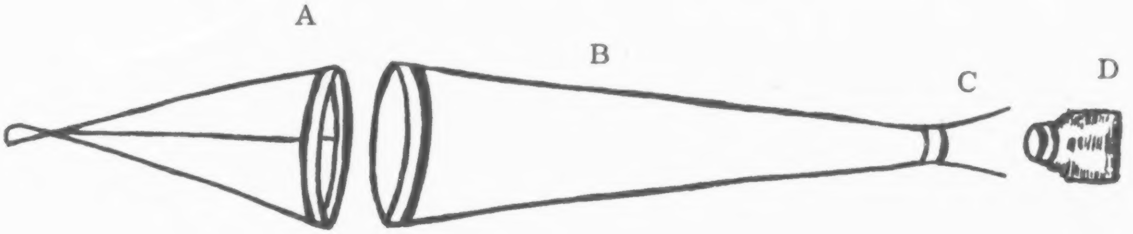

Flat
Raised
Convex
Pulvinate
Umbonate
Entire
Undulate
Lobate
Erose
Filamentous
Curled
Punctiform
Circular
Spindle
Irregular
Filamentous
Rhizoid

QUESTIONS:

1. Did the microorganisms occur in the sea water or the sand?
2. Why would these organisms exist in the sand?
3. Why is each medium of different composition?
4. What function do these organisms have in the marine food chain?
5. At what point (s) in the procedure is contamination most likely to occur?

INVESTIGATION: PLANKTON

BACKGROUND: Since a plankton net is essential for this study, instructions included for its construction:



A. Ring: solidly constructed of brass, steel, with diameter varying from 2 decimeters to 1 meter. Steel wire, chain or rope leaders are attached to the ring and to a single swivel at the forward end.

B. Net: long nylon stockings or silt bolting cloth with about 125-200 meshes to the inch are recommended. Reinforce with canvas around the openings.

C. Ties: sew on two canvas ribbons along the length of the net as shown. The free ends serve to secure the collecting bottle.

D. Bottle: sturdy plastic or glass bottle with deep threads at the neck. Size depends upon net diameter. Plastic mustard and ketchup dispensers, glass cherry, olive or babyfood jars, etc. are adequate.

Biological supply houses generally stock an assortment of plankton nets and accessories. Adapters are available to be permanently fastened into the net opening so that collecting bottles can be screwed into place rather than tied. But, best results will be achieved with the smallest mesh size, whether the net is purchased or constructed.

This particular activity centers around the collection and use of a constructed sample which can then be diluted and distributed to an entire class. If time is available the following types of comparative samples can easily be substituted:

1. Individual samples from the same area
2. Samples from different areas.
3. Samples taken at different times during the day (with reference to light and tide).
4. Samples taken at different depths (vertical sampling).

Pulling the net behind a slowly moving boat is one standard method of collecting plankton. For surface sampling the net should tow just below the surface. For deeper sampling tie a weight to a separate line and fasten to the front swivel. Even throwing the net from a bridge or a pier into a fast tidal current often produces excellent results. The sample needed for this laboratory should be taken in the early morning before school . . . to be used the same day. This plan will almost assure students the rare opportunity to observe living plankton. Place sample in a larger jar or bucket and aerate gently. Dilute with a known quantity of filtered sea water. When ready to begin the exercise, divide the sample into equal portions. Make available the following information to students:

a. date and general location of sampling 10/21, n. Tampa Bay
b. time of day - 0600 hours

c. wave activity and temperature (optional) - light chop 24 C

d. tide - low

e. depth of sample - surface

f. tow time (15 minutes is adequate for a good sample)
15 minutes

g. mesh size - 125/inch

h. diameter of net opening (in meters) - . 2

i. estimated distance net towed (in meters) - 1500

j. dilution - 2:1

Three periods are needed to complete all phases of the exercise. Each day includes a separate purpose, set of procedures and observation guides.

First Day: Observation of living plankton: student should become aware of (a) dominant forms, (b) methods of locomotion, (c) variation in shapes.

Second Day: Measuring and classifying plankton: (a) obtaining representative measurement of dominant forms, (b) identifying common types.

Third Day: Total sample analysis: (a) learning how to count plankton, (b) estimating size of population in comparison to total volume of water sampled.

There exists in the oceans of the world, in seas, bays, lakes, and in nearly every other natural water body, a population of organisms so immense that it defies counting! Everyone, while swimming, has probably brushed up against millions of these creatures without being aware of their presence. No doubt some have swallowed a large number while learning to keep their mouths closed underwater!

Although limited studies of this population were made before 1887, it was not until that year when the oceanographer, Victor Hensen, first proposed a name for this vast assemblage, plankton. The term refers to those plants and animals mostly microscopic in size, that are made "to wander or drift" (Hardy, 1961), under the influence of ocean currents and tides. Even though many planktonic forms have the ability to swim, their efforts in the presence of oceanic water movements are generally too feeble and in vain. Animal members are termed zooplankton. With the exception of marine mammals and reptiles, nearly every creature in the sea spends either a part or all of its life drifting about. Eggs, larvae, and juveniles of most invertebrates and fishes and even some adult forms are common. Copepods (crustaceans) are the most abundant and universally distributed plankters.

Phytoplankton (plants) are more numerous than their animal counterparts, and are best represented by the microscopic diatoms that form the vast bulk of the ocean's vegetation.

Because of their numbers, wide distribution, and beneficial biological activities, plankton are considered the most important inhabitants of the marine world with all forms of life directly or indirectly dependent upon them. Plankton are basic to the food chains of all marine life. Sponges, tube worms, clams, and sea squirts filter out sea water to gether them. Herring catch (Russell and Yonge, 1963) them. Giant baleen whales, over

100 feet in length and reaching fantastic weights of 150 tons, feed exclusively on plankton (Pequegnat, 1958). Diatoms are an important source of vital oxygen and proteins which animal life cannot synthesize but require. Without plankton the seas would surely be a wet desert!

A plankton net with a measured net opening was towed behind a boat for a measured distance. The total number of organisms collected from this tow represents the concentration of plankton in that volume of water.

a. To compute water volume which passed through net towed behind a boat:

$$M^3 = \frac{\pi D^2}{4} \times L$$

M^3 = volume of water in cubic meters
D = diameter of net in meters
L = length of tow in meters

for example a net with a diameter of 20 centimeters was towed 1500 meters,

$$= \frac{3.14 \times .2^2}{4} \times 1500$$

= 47.1 cubic meters

b. To compute volume of water which passed through net tossed from a bridge:

First determine length of tow:
L = length of tow in meters

$$L = \frac{Ti}{t} \times W$$

Ti = length of time net was immersed in water
t = time floating object took to pass width of bridge
W = width of bridge in meters

for example: a net strained water for 18 minutes. It took a cork .25 minutes (15 seconds) to pass under a bridge 12 meters wide. Thus

$$L = \frac{18}{.25} \text{ x } 12$$

$$L = 864 \text{ meters}$$

. . . now compute water volume (assuming that net diameter is still .2 meters).

$$M^3 = \frac{\pi D^2}{4} \text{ x } 864$$

$$= 27.1 \text{ cubic meters}$$

3. Compute the density of the original concentrated sample (number of microscopic specimens/ ________________ml).

The original sample contained __________ml of liquid. It was diluted with an equal volume of sea water. You then received __________ ml of this diluted sample. How many microscopic (visible) organisms are contained in: (a) your sample and; (b) the original sample. Diatoms and other microscopic forms have been eliminated from this count.

a. Pour the sample into a petri dish.
b. Determine the area of the dish.

$$A\ (cm^2) = \frac{\pi D^2}{4}$$

A = number of sq. centimeters covered by dish.

c. Place petri dish over a centimeter grid or graph paper with centimeter squares clearly marked. Distribute sample evenly over bottom of dish.

d. Select, at random, five (5) squares and count all macroscopic organisms in each. To determine average divide total number of organisms by 5.

e To estimate total number of organisms in the petri dish, (thus in the sample):

$$T = \frac{t}{8} \text{ x } A$$

T = total microscopic organisms in the sample

t = total count from random grids

g = number of grids counted

A = area, in cm^2 of petri dish

for example: 100 organisms were counted from 5 random grids. The area of the petri dish covered 78. 5 cm^2.

thus = $\frac{100}{5} \times 78.5$

= 1570 macroscopic organisms/

__________________ml diluted sample

f. Compute total of microscopic organisms in original concentrated sample:

$$Q = T \times \frac{V}{v}$$

where . . .

Q = total misroscopic organism passing through plankton net.

PURPOSE: The purpose is to observe and analyze a plankton sample taken

from ________________.
location

MATERIALS:
- Compound microscope
- Dissecting scope
- Petri dish
- Preservative (3-5% formalin)
- Centimeter grid or graph paper
- Sample bottle and label
- Plastic metric ruler
- Eye dropper
- Standard slides and cover slips
- Concavity slides
- Diluted plankton sample for each item

PROCEDURE:

Part I - To observe living plankton

1. The teacher will provide the recorded conditions under which the plankton sample was made. Enter this information on the data sheet provided.

2. Obtain diluted portion of the plankton sample and pour into petri dish. Gently swirl the contents to distribute.

3. Observe with a dissecting microscope. Scan the entire field.

4. Many organisms are too small to be seen with the dissecting microscope. Prepare wet mounts of the sample and observe under a compound microscope with both low and high power objectives. Do not discard any part of the sample. Empty wet mounts back into the petri dish. Rinse with medicine dropper of water.

5. Do not record any observations today. Look for:

   a. most abundant organisms

   b. variations in shape, color and swimming abilities

   c. types of appendages

   d. chlorophyll-containing organisms

   e. eggs

   f. larval and juvenile forms of crustaceans and fish
   (see pictorial guide to the plankton, pages 68-71).

6. Preserve sample in 3-5% formalin before leaving. Label sample bottle.

Second Day ______________________
Date

Part II - To draw, measure and record characteristics of dominant organisms in plankton sample.

1. Select the most common organism from the preserved sample. Prepare a wet mount and view with low power (or high power).

2. Record the following information on data sheet:

   a. a detailed penciled drawing of the specimen, with labels

   b. measured actual size, in microns

   c. measured drawing size, in microns

d. magnification of drawing

e. identification

3. Repeat this procedure with as many different specimens as time permits. Record all observations.

4. Do not discard any portion of the sample. Empty wet mounts back into sample bottle. Rinse with dropper of water.

Third Day ______________________________
Date

Part III - To complete drawings, measurements and observations on preserved plankton and compute the size of the plankton population in comparison to total volume of water strained.

1. Complete additional drawings, measurements and observations as required.
2. Compute total volume of water strained.

QUESTIONS:

1. How are organisms which are not part of the plankton population classified?
2. What is the DSL?
3. Which environmental factors may influence the vertical migrations of plankton?

T = total number of organisms in your sample.

V = total volume of original concentrated sample.

v = Volume of your sample

for example: Ten (10 ml) of sample was strained from 30 cubic meters (M^3) of sea water. You received 1 ml of this and proceeded to count 1570 microscopic organisms. Thus

$$\frac{1570 \times 10}{1} = 15.700 \text{ microscopic organisms/}$$

$30M^3$g. Compute the total number of microscopic organisms per cubic meter.

4. How do the minute and delicate plankton withstand the crushing pressures of deeper waters?

5. How do diatoms and copepods enter into the food chains of marine organisms?

6. How do baleen whales feed on plankton?

7. Are there ocean areas in the world devoid of plankton?

8. What was your impression of the abundance of diatoms in the plankton sample? Copepods?

9. Which phyla of organisms were best represented in your sample?

10. What happens to plankton which "drift" into waters where conditions of salinity, oxygen, or temperature are unfavorable? Explain your answer.

11. What are some limitations to using a plankton net in sampling a population or organisms?

12. What are some limitations to the methods used in counting the density of plankton populations? What are other methods of estimating the density of plankton?

13. What is meant by "standing crop of plankton"?

14. What do the prefixes, "holo-, mero- and nano-" refer to in reference to plankton? Give examples.

15. What adaptations do plankton have for moving?

INVESTIGATION: PHYTOPLANKTON & PROTOZOA POPULATIONS

BACKGROUND: All living things can be classified on the basis of their nutrition into one of two groups; the heterotrophs and the autotrophs. The heterotrophs require one or more organic nutrients; they are said to have an animal-like nutrition. The autotrophs, on the other hand, do not require organic nutrients and subsist upon soluble inorganic compounds instead. Algae are photosynthetic autotrophs that use CO_2 in place of organic compounds as a source carbon. Protozoa are heterotrophs, since they "eat" living or dead organic matter. Protozoa and algae make up a large portion of the free-floating aquatic organisms referred to as plankton. The algae are phytoplankton and the protozoa are zooplankton.

The numbers and kinds of algae in natural waters are one measure of the health of that body of water. If too few algae are present there will be little production of aquatic animal life, since algae form the basis of the food web. Too many algae might indicate that there is an excess of phosphate or nitrates in the water. The processes resulting from such over enrichment is called eutrophication.

A careful count of the algae at a study site might indicate nutrient pollution or simply a highly productive environment. The specific kinds of algae or protozoa present can also be indicators of aquatic conditions. For example, an abundance of nitrogen-fixing blue-green algae might indicate a shortage of available nitrogen from sources other than the atmosphere.

PURPOSE: To determine the numbers of protozoa or algae in fresh or salt waters.

MATERIALS:

Micropipettes 0.01 ml capacity
Cover glasses, 18mm x 18mm
Microscope slides
Centrifuge tubes, 15 ml, graduated at 1 ml
Centrifuge
Aspirators

Optional: Colorimeter (Spectronic 20)
Alcohol, hot plate, beaker

PROCEDURE:

1. Shake sample to mix and measure 15 ml. portions in graduated

centrifuge tubes. Centrifuge 5 min. at 3000 rpm.

2. Using an aspirator, draw off the supernatant leaving 1.0 ml. containing the plankton in each tube.

3. In preparation for counting, shake the tube to mix and place a drop of the fluid on a slide using the 0.01 ml pipette. Top with a cover glass. A drop from the duplicate tube may be placed on the same slide. Using the 43x objective, count a strip from one edge of the cover glass to the other and a strip from top to bottom.

4. Tabulate the blue-green algae, green algae, and diatoms separately.

5. Calculation -

The cover slip area consists of 160 fields, 40 across and 40 down.

$\frac{80}{1600}$ of the area was counted. 0.01 ml was used. This was 15 times concentrated.

$$\begin{array}{c} \text{No. plankton} \\ \text{per ml} \end{array} - \begin{array}{c} \text{No. plankton} \\ \text{counted} \end{array} \times \frac{20}{1} \times \frac{100}{1} \times \frac{1}{15}$$

$$'' \quad - \begin{array}{c} \text{No. plankton} \\ \text{counted} \end{array} \times 133/\text{ml}$$

If there are too many plankton per field to count easily, add an aliquot of water to centrifuge tube and adjust calculation accordingly.

OPTIONAL: The total photosynthetic potential of a water sample can be estimated by extracting the chlorophylls and other pigments from the algae and quantitating their absorbance in a colorimeter.

PROCEDURE:

1. Centrifuge and aspirate the water sample as described above.

2. Add 5 ml of alcohol to the pelleted algae. Mark the tube at 5 ml.

3. Place the tube in a boiling water bath for about 7 minutes in order to dissolve the pigments. Be careful not to boil away the alcohol -- add additional alcohol so the level remains at 5 ml.

4. Centrifuge at 5000 rpm for 5 min. to remove dead cells and precipitates.

5. Pour the contents into the colorimeter cuvette set at a wavelength in either of the two chlorophyll absorption peaks, e.g. blue-violet or red.

6. Dilute the sample with more alcohol if the optical density is greater than 0.4.

Dilution Factor (D. F.)

$$= \frac{\text{amount of chlorophyll solution + amount of alcohol added in mls.}}{\text{amount of chlorophyll solution}}$$

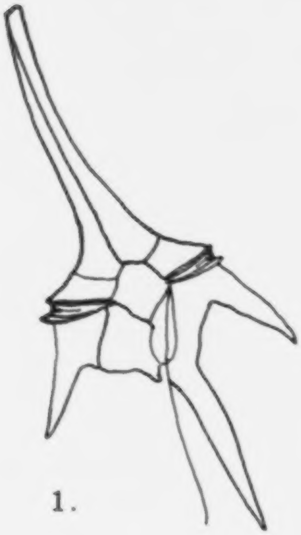
D. F. x optical density (O. D.) = total O. D.

7. Data should be presented in a tabular or graphic manner. Label all units of measure and include a legend describing the methods unique to this data. Express data in optical density (O. D.) units at a particular wavelength in millimicrons.

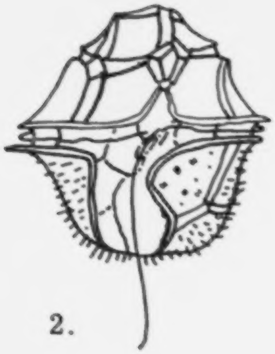
REFERENCES: The cell count method described here is adapted from the one presently used by the Florida State Board of Health at Rockledge.

Clean Water Applied Biology, H. M. Freeman and Co.

Colorimetric Procedures and Chemicals for Water and Wastewater Analysis, 4th Ed 1970.



1.



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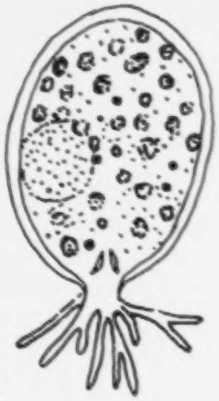

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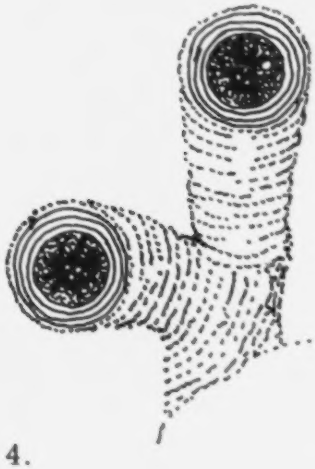

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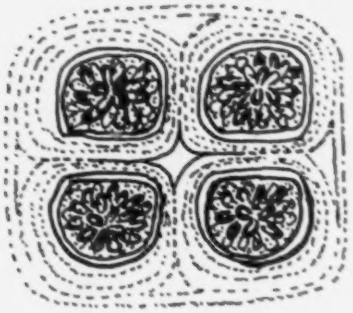
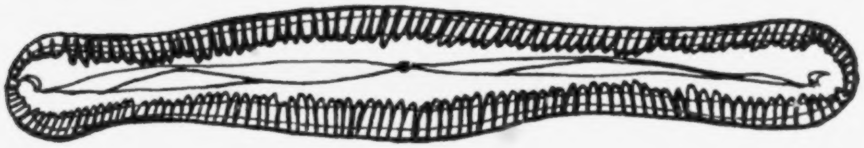


5.

1. Ceratium hirundinella
2. Peridinium palatinum
3. Codinilm limnetcon
4. Urococcus insignis
5. Gloedinium montanum



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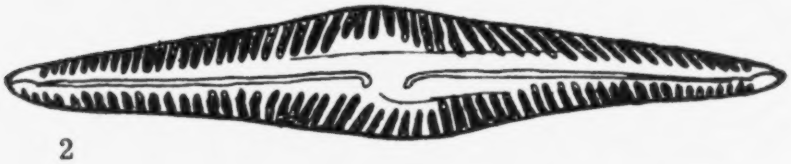

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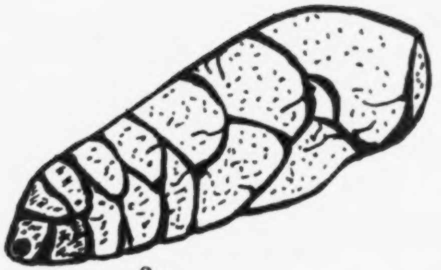

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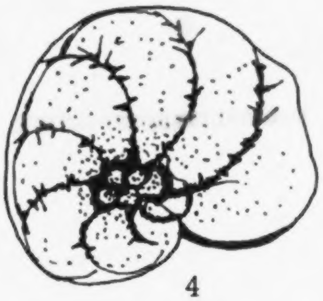
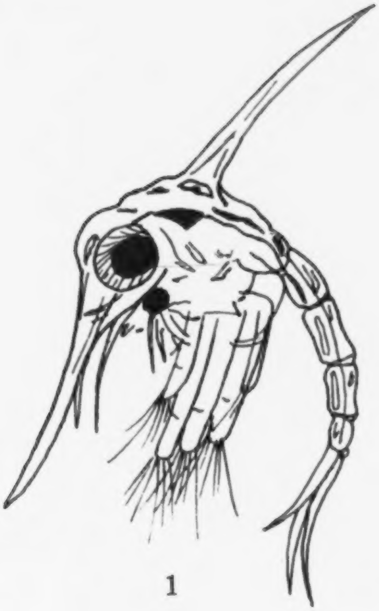


4

1. Naviculoid diatom, Pinnularia nobilis
2. Naviculoid diatom, (Chrysophyta)
3. Foramenifera, Globerigina sp.
4. Tintinnoidea, X (Protozoa)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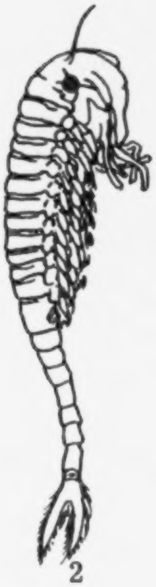
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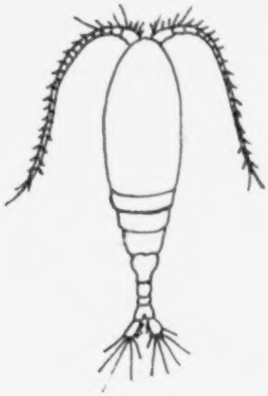
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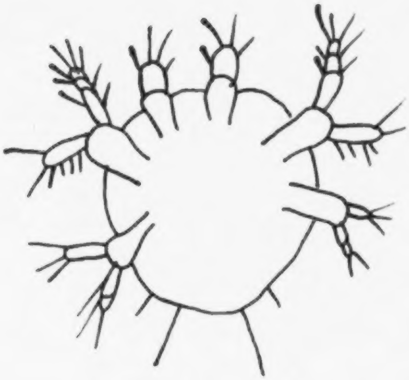
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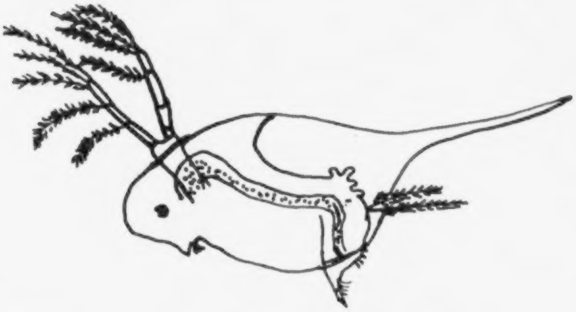
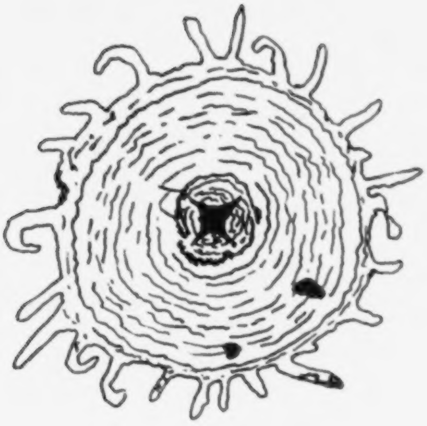

5

1. Zoea larva of crab (Arthropoda)
2. Amphipod (Arthropoda)
3. Copepod (Arthropoda)
4. Nauplius form of copepod (Arthropoda)
5. Phyllosoma of spiny lobster (Arthropoda)



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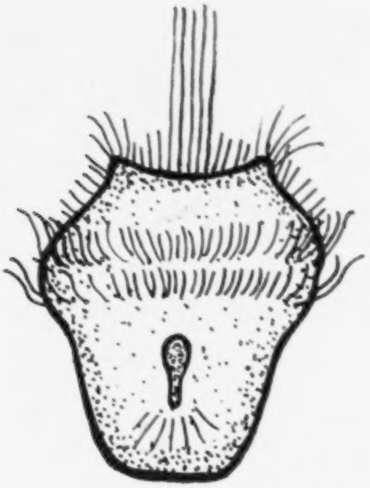

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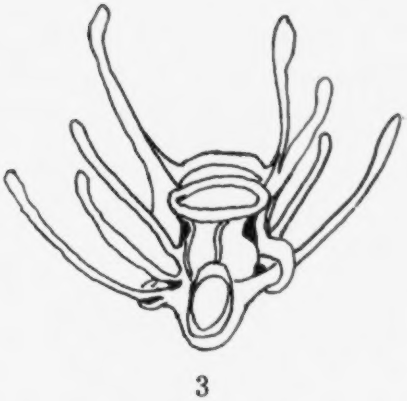

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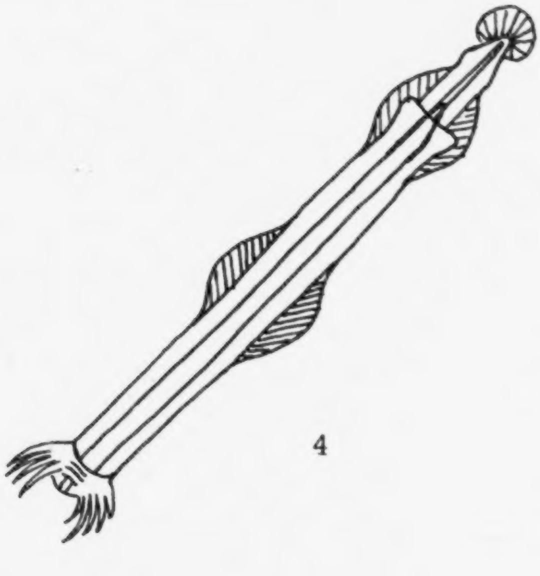


4

1. Medusa of the hydrozoan, *Obelia sp.*
2. Veliger larva of a gastropod
3. Pluteus larva of a brittle star
4. Arrow worm, *Sagitta sp.*

PLANKTON LABORATORY

DATA SHEET

I. Record the following conditions pertaining to the sample under study:

a. date ____________

b. location ____________

c. time of day ____________

d. tide ____________

e. depth ____________

f. tow time ____________

g. mesh size ____________

h. diam. net opening _____ meters

i. distance net towed _____ meters

j. dilution of your sample ____________

k. volume of your sample _________ ml

II.

organism #	1	2	3	4	5	6
drawing						
actual size						
drawing size						
magnification						
identification outstanding features ? coloration ? common and/or scientific name						

III. a. volume of water which passed through the plankton net ____________________ M3

b. number of macroscopic organisms in your sample (T) ____________________

c. number of macroscopic organisms in original sample (Q) ____________________ M3

THE NATURAL HISTORY OF MOSQUITOES

Maurice W. Provost

Mosquitoes may appear to be pretty well known insects, yet almost half the literature on them relates to two domestic, disease-carrying species, Aedes aegypti and Culex pipiens, and the bulk of the remainder relates to those domestic species of Anopheles which carry malaria in one part or another of the world. Now, it's understandable that disease-carrying mosquitoes should have received the most attention, but the fact remains that these domestic species are simply not representative of the world's 1500 mosquito species. Most mosquitoes are wild, and our big problems in Florida are mostly with such wild mosquitoes as Aedes taeniorhynchus, Aedes sollicitans, Psorophora confinnis, Culex nigripalpus, and Coquilletidia perturbans. In our researches here we are forever having to overcome time-honored ideas about mosquitoes which may be true of domestic forms but simply cannot apply to our wild species.

When we speak of domestic and wild mosquitoes we are grouping them in broad categories which can be broken down further. For instance we can divide the wild ones into woodland and field species, and these further into swamp mosquitoes, savannah mosquitoes, jungle mosquitoes, tundra mosquitoes, and so on. These would be ecological groupings. Obviously, you can group mosquitoes all kinds of ways, depending on your criteria. If you're concerned with their biting habits, as epidemiologists have to be, you'll divide them into non-biters, man-feeders, mammal feeders, bird feeders, and so on down the line. If you're interested in what time of day they fly, as mosquito-control workers are bound to be, you'll speak of nocturnal species, diurnal ones, crequscular or twilight fliers, and so on. But it is very important to remember that the most basic grouping of all is the one you'll learn under Dr. Pratt and his staff, viz. the classification based on anatomy, or structure. This is the grouping that leads to scientific names, and unless you can name them you can't talk about mosquitoes at all.

My topic is so broad that I could talk forever and bore you to death. I won't talk forever and I'll try not to bore. I'll discuss a few aspects of mosquito natural history where important new findings have been made in recent years and which I think will be of interest to you. As a sort of preface and introduction to Florida's mosquitoes we'll start with a few words on seasons and distribution.

SEASONS AND DISTRIBUTION

For over 20 years now, the Florida list of mosquitoes has stood at 67 species, so I guess that's where it will stay. The whole Southeast has only 7 more, or 74 species. What is worst for Florida is that 25 to 30 of those 67 species are real pests and a good dozen severe pests. The distribution of these 67 species in Florida fits well into what we call biotic areas and which accommodate all forms of life. These areas are based on climate and other ecological factors.

Florida is usually thought of as having two seasons, a hot and wet summer and a cool and dry winter, but it's really not that simple. We have four seasons (SLIDE 1) based on temperature: 3 months each of winter and spring, 4 months of summer, and 2 months of fall. Everyone knows that in the winter north Florida is colder than south Florida and that there's not much difference in the summer. It's best, however, to look at temperature extremes and not just at averages. Thus (SLIDE 2) there's a marked difference in cold nights per year not only between Tallahassee and Key West but between Orlando and Ft. Pierce. And in the summer (SLIDE 3), when mean temperatures show so little geographical difference, there are great differences in numbers of very hot days, and north Florida is much hotter than south Florida. As for rain, we can say most of Florida averages 50 to 55 inches a year, with a little more in the western panhandle and the southeast coast and a lot less in the Florida Keys. The proportion of the year's rain (SLIDE 4) falling in the wet half of the year increases from 55% in west Florida to over 80% in southwest Florida. So the "wet season" is a phenomenan of peninsular Florida. The mosquito-breeding season slackens in the winter because of lack of rain in south Florida and because of cold in north Florida.

Our Florida mosquitoes breed mainly in the summer, but we have exceptions. At least 3 species are winter breeders: Anopheles punctipennis, Culex restuans, and Culesita inornata. The famous one-generation-a-year mosquitoes of the far North are represented in Florida by only one species, Aedes sticticus. It comes off the river flood-plains of north and west Florida in the spring, along with Aedes vexans, infirmatus and fulvus-pallens and Psorophora discolor, varipes and ferox. Another seasonal oddity in Florida is Culex salinarius; it breeds all year but reaches peak numbers in late spring, just ahead of the rainy season. And, finally, a word should be said about a serious pest, Coquilletidia (once Mansonia) perturbans. Throughout North America this is a one-generation-a year species, overwintering as fourth-instar larvae. In Florida we frequently have peaks of emergence in both spring and fall or even in fall alone. We are trying to find out whether this means a second generation a year or alternating spring and fall broods.

Biotically, Florida straddles the line dividing (SLIDE 5) the Western Hemisphere into a Nearctic Region extending to the North Pole and a Neotropical Region reaching to the South Pole. Mainland Florida falls into what biogeographers call the Austroriparian life zone, which in the State can be further divided into biotic areas labeled Subtropical, Floridian, and Louisianian. Twenty-seven mosquito species (SLIDE 6) occur throughout Florida. Among the species which have either their northern or their southern limits of distribution in Florida (SLIDES 7-10), slightly more come from the North than from the Tropics. (See attached maps).

GROWTH AND DEVELOPMENT

The emergence of the adult (SLIDE 11), from the pupal skin and from the water, ushers the mosquito into a life as different from what went on before as any two forms of life could possibly be. I will talk mostly about the adult, but first I will discuss some of our findings about how long it takes the growing mosquito, which is the larva, to reach adulthood.

Growth rate depends on four factors: temperature, food, water chemistry (particularly salinity) and larval density. In the laboratory we have measured the separate effect of each of these factors in 17 species of Florida mosquitoes. In seven of these, *Aedes taeniorhynchus* and *sollicitans*, *Culex quinquefasciatus*, *nigripalpus* and *bahamensis*, *Psorophora confinnis*, and *Anopheles crucians*, we found that the time of day the eggs hatched was a fifth factor because the change from fourth instar larva to pupa came at a certain time of the day only. Thus (SLIDE 12) *Aedes taeniorhynchus* pupates in the afternoon, no matter when it hatched, so that the ones ready to pupate shortly after sunset, for instance, put off actual pupation 18 hours or until the afternoon of the next day, while the ones ready to pupate in mid-afternoon go ahead and pupate right then and there. The ten species we found out had no such pupation rhythm but pupated as soon as developmentally ready, regardless of the clock, were *Aedes aegypti*, *vexans*, *infirmatus* and *triseriatus*, *Psorophora ferox*, *Anopheles quadrimaculatus*, *Culex salinarius*, *Deinocerites cancer*, *Culiseta melanura* and *Wyeomyia vanduzeei*.

We also studies the effect of all these growth factors on the kind of adult produced. Certain basic sexual and species differences are never changed. In the broods or populations, females of *Aedes taeniorhynchus* will always be larger and heavier than males, whereas in *Culex nigripalpus* the sexes will be alike in size and weight. Also, *Aedes aegypti* and *triseriatus* will always emerge fatter than other species similarly reared and have therefore a longer inherent life if starved as adults. On the other hand species vary in the amount of flight-energy researve, especially glycogen, they emerge with. *Aedes taeniorhynchus* and *sollicitans* and *Psorophora confinnis* have a lot more than other mosquitoes, while *Aedes triseriatus* and *Psorophora ferox* have very little. And so it is that, when it comes to longevity and flight potential, certain species of mosquito have the jump on others from the very moment they emerge as adults.

LONGEVITY

Although we may observe that broods of Glades mosquitoes last about 10 days and broods of Salt-marsh Mosquitoes about three weeks, individual mosquitoes don't average that long a life. Investigations throughout the world have shown mosquitoes to be pretty short-lived animals. I remember when it was announced in a tropical medicine journal that the most important malaria mosquito in India, *Anopheles culicifacies*, lived on an average only four days, or, to put it another way, females died off at a rate of 50% every other day. This was 29 years ago, but malaria workers had known for years that even when the female picked up the infection on her first bite, the incubation period required her to be at least two weeks old before she could transmit malaria to another person. So an important point in population dynamics was established: even with 50% dying every other day, enough females lived over two weeks to keep malaria propagating at one of the highest rates in the world.

A lot of work has been done on mosquito longevity in the past three decades and it can be summed up this way. All species studied (SLIDE 13) have female mortality rates in nature somewhere between 40% and 85% every two days, i. e. they live on an average 3 1/2 to 16 days. Our own studies give females of *Aedes taeniorhynchus* and *Culex nigripalpus* average lives of 6 and 4 days respectively. In these as in most, if not all, species the males live shorter lives than females.

If we now go to the laboratory with *Culex nigripalpus*, we learn (SLIDE 14) that females starved from emergence live at the most only 5 days. The survival curve is shaped differently here because we've eliminated all the things that kill adults in nature except one: starvation. If they're fed sugar they live on and on; one lived 98 days. This year we've shown that *Aedes taeniorhynchus* females on sugar alone live much longer than those on blood alone and longer even than those on blood and sugar. I should emphasize at this point that how long mosquitoes live when starved from emergence is a special characteristic. In *Culex nigripalpus* 50% of the females die in 2 1/2 days, in *Aedes sollicitans* 50% die in 5 days, in *Aedes aegypti* it's about 12 days, and in *Opifec fuscus*, a New Zealand mosquito we've worked with here, it's around 18 days. These differences reflect differences in how much fat reserves the females emerge with, as I've indicated before. *Opifex* females emerge as veritable butterballs, but *nigripalpus* females always emerge lean and hungry. This leads us straight into our next subject.

ADULT NUTRITION

For years the notion was around that female mosquitoes fed on blood and males on flower nectar. More than 20 years ago we had seen so many females of so many species feeding at flowers and extra-floral nectaries that we were convinced sugar was the food of both sexes. Now, after years of hard work in the chemical laboratory we know pretty much what the story is. Females use blood to make eggs, which was known long before our time, but for energy purposes they get very little out of blood. In order to live long and to fly any appreciable amount they must eat sugar.

There is still a tremendous difference between the sexes, however, but it's not what was once thought. The male (SLIDE 15) cannot convert sugar into fat while the female can and does. Both sexes convert sugar into glycogen, or animal starch, but only up to a fixed and small quantity. What we need to remember is that in order to fly, mosquitoes must have sugar or glycogen for fuel. Once these energy reserves are used up they can't fly anymore, --no matter how much fat is left in them . Fat is used for survival energy, or basic matabolism, so the more there is the longer they can live. Since the male cannot manufacture fat, he survives on sugar and glycogen. And since he can store only a small quantity of glycogen he must keep up his reserves of sugar by feeding on sweets almost every day. This, of course, explains why male mosquitoes are so often seen on flowers. The female (SLIDE 16), however, turns a lot of sugar into fat, and after one really good nectar meal she can live for days or even weeks off the fat she's stored. And this is why female mosquitoes aren't seen at flowers as much as males.

Should you wonder how these energy researves and conversions are measured, I can say only that it couldn't be done until we had developed very accurate micro-analytical chemical procedures. Mosquitoes on the verge of dying from starvation could then be proven to be out of sugar, glycogen and fat. At that moment a measured amount of food could be given them and then later traced chemically as it was burned up or converted into some energy reserve or other. The quantity of blood (SLIDE 17) taken was the increase in the mosquito's weight (SLIDE 18) after feeding. Sugar solutions fed could be measured (SLIDE 19) very precisely because a micro-pitette was used (SLIDE 20) and the meal was calibrated in advance. In other words, they could be force-fed sugar but not blood.

I've implied earlier that only the very old female mosquitoes can transmit malaria. Actually all disease organisms carried by mosquitoes have a long incubation period in the mosquito, 10 days being a little short of average. If you add an average two days before the first bite (SLIDE 21), the female must be 12 days old, then, to transmit a disease. Going back to our survival curves we can see that in ordinary broods only 7 1/2% of Aedes taeniorhynchus would live long enough to transmit diseases and only 1 1/2% of Culex nigripalpus would. It's clear from this that whether or not mosquito females get at sugar early in life has a great deal to do with whether or not they live long enough to be disease vectors. The only exceptions would be those domestic species, like Aedes aegypti, which happen to emerge with lost of fat to survive on and which need to do little flying to get at blood, shelter and oviposition sites. These, which are exceptions among the world's 1500 mosquito species, might be able to get what little supplemental energy reserves they require out of blood alone.

MATING AND DISPERSAL

Six years ago it was first announced that in *Aedes aegypti* only one male's sperm got inside a female. A year later it was announced that the male's accessory-gland fluid once in a female prevented her from ever again accepting sperm. We entered the act by announcing in 1968 that the female *Aedes aegypti* could be inseminated only at an age determined by the hormone from her corporata alata, even though she copulated repeatedly before and with every appearance of success.

These findings left us thoroughly confused. In the late forties several of us had observed and reported mass mating on the migratory exodus of *Aedes taeniofhynchus* when the females were only 8 to 18 hours old. Had we seen only sexual play? And females of this and all other species were so invariably inseminated when collected in nature that we just assumed that mating and sperm transfer occured soon after emergence in virtually all mosquitoes. Now we had to prove or disprove this hypothesis.

Starting first in the laboratory, we learned that females of *Culex nigripalpus* and *quinquesfasciatus* and *Aedes taeniorhynchus* behaved exactly like *Aedes aegypti*. Then came the difficult task of finding out what happens in nature. This involved releasing millions of marked mosquitoes of known age, which was done in 1969 with *Culex nigripalpus* and *quinquesfasciatus* and this past spring and summer with *Aedes taeniorhynchus*. We are still working over the results, but it appears certain that in these three important Florida mosquitoes the females are not inseminated until their second day of adult life and do not even approach 100% insemination for a brood until their third day.

We used to wonder why most mosquito females are 2 or 3 days old before they first bite, but an explanation is now appearing: they are busy eating (sugar, that is) and mating. If they can get the business of becoming fat and inseminated over with in their first three days of life, it is obvious they can spend the rest of their lives biting and laying eggs. The important point is that they first guarantee two things: (1) they'll live long enough to lay eggs, and (2) those eggs will be fertile.

By dispersal we mean the geographical area occupied eventually by the adult mosquitoes of a population emerging from a certain breeding area. It's a function or end result of all the flights performed by those adults. Unless there is an initial migration these are all searching flights, --searching for nectar, blood, shelter, a place to lay eggs, and so on. If a species like *Aedes* aegypti can find all it needs within a hundred yards, then that is how far a population disperses from the breeding site. If *Anopheles quadrimaculatus* commonly flies hundreds of yards to reach one or more of its biological needs, then a population may well occupy the land within a mile of its point of origin. It should be clear that the dispersal of any population of any species will depend altogether on the distribution over the land in question of what it takes to satisfy its biological needs. A rigid "flight range" for a species is just fantasy.

A word, finally, about migration. In those species with a circadian rhythm of pupation and emergence, the adults emerge in large batches or pulses at a certain time of day or night. Thus, in Aedes taeniorhynchus (SLIDE 22) a brood usually emerges in three days of bursts, with males ahead of females. If they are over 6 hours old at senset, the whole day's emergence will take off about 18 minutes after sunset in a spectacular migratory exodus. In experimental work we allow part of the brood to emerge (SLIDE 23) in special cages from which the sloping lid is later removed so that the mosquitoes can leave at will, i. e. spontaneously. The back of the cage, up which the new mosquitoes crawl, is gridded and is photographed (SLIDE 24) every 2 minutes, using an electronic flash to which we know the mosquitoes are blind. Using mosquito counts on random squares, we can later plot the results (SLIDE 25) and get a very accurate picture of the exodus. Although it's getting dark when this happens, good eyes can easily see the exodus of salt-marsh mosquitoes, and when it involves millions of billions of mosquitoes (SLIDE 26) it is a spectacle never forgotten.

In the laboratory we have recently learned that the females of Aedes taeniorhynchus exhaust all the energy reserves they emerge with in 3 or 4 hours of flying. This is proven by flying them to exhauston on flight mills (SLIDE 27-21) which compute the time and distance flown, after which what little sugar or glycogen remains in them is measured chemically. This was also demonstrated in acoustic pick-up chambers (SLIDES 32-37) where the sound of spontaneous flight is picked up, recorded and analyzed. Recent experiments using the acoustic boxes have furthermore demonstrated that (SLIDE 38) females produced from crowded larvae fly a great deal more in their first two or three nights than females from uncrowded larvae. We have borrowed the terms long used by researchers with migratory locusts and called these the migratory and non-migratory phases of *Aedes taeniorhynchus*.

In closing I will summarize by stating this, that we have learned so many new and surprising things about mosquitoes in the last five years or so that many of us are truly overwhelmed. There is every reason to expect that in a few years we'll be able to assemble all the loose bits of information into a very different and, we hope, very accurate Natural History of Mosquitoes.

FISHERIES BIOLOGY

I. AQUATIC BIOLOGY

A. Community Structure

1. Sun-source of energy. The sun is the basic source of all energy on earth. Sugar, alcohol, starches, proteins, fats and oils are all producers of heat or energy in the body of an organism and come from plant life directly or indirectly. In an aquatic environment the type of plants produced by the sun are algae (simplest) and higher forms such as hyacinths, elodes, cattails, etc. The three basic nutrients required by all plants are nitrogen (N), phosphorus (P) and potassium or potash (K). These nutrients may come from the atmosphere, the bottom, the water or the watershed.

2. Primary producers, algae and aquatic plants.

Plankton - Collectively, all those organisms suspended in the water of an aquatic habitat which are not independent of currents and other water movements. Most such organisms are microscopic in size and include bacteria, protozoans, rotifers, larvae and small crustaceans and algae.

Zooplankton - Microscopic animals suspended in water.

Phytoplankton - Microscopic plants suspended in water. This term is sometimes used to refer to algae, but algae is a more commonly accepted name.

a. Plankters reproduce quickly, have a high nutritive value, are small (microscopic) in size, and therefore available to all fish. They are found in both fresh and salt water.

b. Since algae are plants and most plants contain chlorophyll, algae impart color to water.

c. Aquatic weeds - Can be both beneficial and a nuisance. Some plants remove excessive nutrients from the water and provide shade and cover for fish. Certain types of submerged aquatic weeds are a haven and sanctuary for many species of aquatic organisms upon which fish feed. However, weeds may give small fish a place to hide and make predation by large fish impossible.

3. Primary consumers of algae are rotifers, protozoans, copepods, aquatic insects, gizzard shad, threadfin shad, and some species of whales. All of these are filter feeders and possess the capability of straining water to remove algae. In this case, plant matter is converted to animal tissue.

4. Secondary Consumers.

a. Forage fish, in turn feed upon primary consumers thus converting animal tissue to fish flesh.

5. Tertiary Consumers.

a. Large predators feed on forage fish and convert this into additional fish flesh.

6. When death of secondary and tertiary consumers occur, nutrients (N. P. K.) are returned to the water to carry on the cycle. "Matter can neither be created nor destroyed."

B. Eltonian Pyramid

1. Trophic level - The transfer of food energy from source plants through a series of organisms with repeated eating and being eaten is referred to as a "food-chain." Food chains are of three types.

a. Predator Chain - Plant base to smaller animals to larger animals.

b. Parasite Chain - From larger organisms to smaller organisms.

c. Saprophytic Chain - From dead material into microorganisms.

Food chains are inter-connected with one another and the whole pattern is sopken of as the food web. In natural communities, organisms whose food is obtained from plants by the same number of steps are said to belong to the same "trophic level." Charles Elton, an English ecologist, in the 1920's and 1930's was the first to clarify this concept and use what is known as the Elotian Pyramid to simplify this principle.

The pyramid is used to explain Elton's principle in that it visually illustrates the large amount of a given substance utilized to produce a smaller mass of a higher organism. Sunlight (energy) coupled with carbon dioxide through the process of photosynthesis is converted into plant material plus simple sugar and oxygen. There is a tremendous loss of the energy tied up in sunlight compared with the amount of algae, sugar and oxygen produced. Consequently there is a further loss of energy as phytoplankton is converted into zooplankton to the ultimate consumer--man.

C. Food Web - Food chains are inter-connected with one another to form a food web: in the overall relationship, green plants occupy the first plateau, plant eaters the second, carnivores the fourth level.

D. Expressions of Producitivity

1. Carbon Fixation - Carbon in the form of carbon dioxide and hydrogen from water are combined by energy from light in the process of photosynthesis to produce phytoplankton. The resulting zooplankton density has a direct bearing on fish yield in a given body of water. There are more elements involved in this process other than carbon (nitrogen, phosphorus, potassium) and other factors have an effect, such as pH.

E. Standing crop and its constancy

The standing crop is the weight of all the organisms within a plateau that can be supported by a steady and constant flow of energy in a food chain. It varies in accordance with the amount and size of the organisms available.

F. Population dynamics and ecological relationship

Another way to express this heading is to refer to the changes in a fish population in relation to their environment (the lake).

1. Goldfish not laying eggs at high population density. This phenomenon can occur in any organism, but man does not always understand why. In the case of goldfish, a suppressive factor is involved which inhibits reproduction when the population density reaches a certain high point.

2. Oyster drill - oyster relationship. The oyster drill is a small (one inch) marine snail with a shell having a ridged surface and a scalloped lip. It destroys oysters by drilling a small hole in the shell and sucking out the juices by means of a long proboscis. The density of oyster drills in a given area has a direct relationship to oyster production.

3. Oyster - algae relationship. Great South Bay in Long Island Sound, N. Y. provides a dramatic example of how too much of a good thing can completely change an ecosystem. Large duck farms were established along the tributaries leading into the bay and resulted in extensive fertilization of the water by duck manure and a consequent massive increase in phytoplankton

density (the low circulating rate in the bay allowed the nutrients to accumulate rather than be flushed out to sea). This produced a complete change in the type of phytoplankton. The mixed population of diatoms, green flagellates and dinoflagellates was replaced by a green flagellate of little known origin. The famous "blue-point" oysters of that area were unable to utilize the newcomers as food and gradually disappeared.

4. Bass-bluegill relationship; tendency to go to many small fish. The best way to describe the bass-bluegill relationship is to say there is a "balance" between the predators (bass) and forage fish (bluegill). Too many bass result in the bluegill being eliminated and stunting of the subsequent bass generations. Too many bluegills inhibits bass reproduction and results in subsequent bluegill generations being stunted because of over-crowding. Sportsmen prefer to catch bass more so than bluegills and can be responsible for tipping the "balance" in favor of the bluegill by catching too many predator fish. Bluegills also have a higher reproductive capacity than bass; however, bass require many pounds of bluegill to produce one pound of bass.

II. THE ENVIRONMENT

A. Physical-chemical characteristics and effects

1. Oxygen - limiting effect on fish kills. The most common type of fish kills are those resulting from oxygen depletion due to

algal die-off. The severity of the kill is directly related to the amount of oxygen removed from the water by decomposing algae. The critical level is 3 ppm. Below this, game fish begin to die. The critical period is just at dawn when reverse photosynthesis has been occurring all night and phytoplankton has been producing carbon dioxide. With the oxygen demand made by decomposing algae, this whole process results in an ocygen depletion that triggers a fish kill until the oxygen is replaced, usually by photosynthesis later in the day.

Fish kills occur more often in warm water than cold because the warmer the water, the less oxygen it can absorb.

2. pH - This is a measure of the acidity or alkalinity of water. Generally speaking, water that is neutral (neither acid or alkaline) is the best producer of fish. On a numerical scale from 1 to 14, below 7 is acidic, above 7 is alkaline and 7, obviously, is neutral.

3. Temperature: tolerance range and acclimation. The tolerance range for each specie of fresh water fish in Florida is not known. So called warm-water fishes can live indefinitely at low temperatures to which they are normally exposed in winter, but do not thrive in waters which remain cold throughout the year. As a result of depressed metabolic rate, they are not active and grow little or not at all at the low temperatures, and successful spawning and development may be impossible.

Conversely, cold water fishes, such as trout, may be unable to compete successfully with warm-water species in environments in which summer temperatures are higher and dissolved oxygen concentrations low, though not lethal. The highest temperatures to which different species can be gradually acclimatized has been found to be about 105.8° for goldfish. Each species of fish has a different maximum temperature to which it can become acclimated.

B. Evolution of a lake to terrestrial environment

1. Oligotrophic - May have been formed by glacier, volcanic upheaval, or meteor. Characterized by deep water, little aquatic vegetation, low-temperature and cold water species of fish (trout, pike, salmon).

2. Eutrophic - Erosion may have started resulting in shallow banks, higher water temperatures, increased aquatic plants around margin and warm water species of fish (bass-bluegill).

3. Dystrophic - More silting and erosion occurs, hence, there is less deep water. Bottom sediments accumulate. Temperatures rise, algae predominate. Fish species change to gar, shad and catfish.

4. Terrestrial

   a. Marsh - No deep water. Rooted aquatic plants dominate. Few or no fish except minnows.

   b. Swamp - Characteristics similar to marsh environment, no deep water, aquatic plants, and trees dominant forms.

c. Lowland Forest - No visable water or aquatic vegetation. No fish. Trees or brush area of a type that require wet or damp ground for maximum growth.

III. FISHERIES MANAGEMENT

A. Stock those species which will best be adapted to the environment.

In most cases this means culturing and propatating native fish such as bass, bluegill, shellcracker, and channel catfish.

B. Study yield - Increase fertility.

Yield can best be increased in small ponds by periodic fertilizing with inorganic materials and/or supplemental feeding.

C. Thinning

In a bass-bluegill combination farm pond, it is usually best to destroy all forage fish caught with hook and line regardless of their size. The more pounds removed, the larger fish remaining will grow to the carrying capacity of the lake.

D. Trapping, netting, shocking, poisoning

If a lake is not fished heavily enough with hook and line, more drastic measures of fish removal are sometimes necessary in order to achieve balance. All of the above mentioned headings are designed to prevent forage fish from over-populating themselves to the extent that most of the fish are stunted.

E. Improving Habitat

1. Stream improvement projects. Florida has few swift flowing streams that can be improved for fish management. Baffles or reflectors are sometimes constructed of rock material to further aerate the water. Pools may be interspersed throughout a stream to create more "space" for fish. This can be accomplished with the introduction of beaver colonies.

2. Brush piles in ponds and lakes. These measures have a tendency to concentrate fish, especially where they are fed in the vicinity of such attractions. Florida has used conduit pipe, concrete blocks, tires, large diameter rock and tree tops to construct brush piles. The principle behind the theory that brush piles attract fish stems from the fact that invertebrate fish food organisms attact themselves to the reef material and thereby provide food for forage fish. Forage fish in turn attract predator fish. Brush piles are most effective in large lakes with little marginal cover.

3. Nail kegs for catfish. The channel catfish is the most desirable of the catrishes to the angler. They will not reproduce under natural conditions unless they can spawn in a cavity. In nature they spawn in a hole in the bank of a stream, lake, or in hollow logs. Fishery biologists can compensate for this sometimes lack of suitable spawning sites by depositing nail kegs, conduit pipe and drums in waters to provide the necessary "cave".

4. Controlling aquatic plants. As with brush piles, there is a definite need for limited aquatic plant growth in large lakes, especially around the margin. If limited to the margin, they tend to concentrate fish and remove excessive amounts of nutrients from the water that could otherwise stimulate the growth of undesirable blue-green algae. Certain aquatic plants provide a haven and support structure for various species of insects and other invertebrates.

Unlimited plant growth throughout the lake hinders fishing, boating and water skiing. These other aspects of multiple use of recreation areas must be considered in the long range overall concept of fisheries management.

5. Destroying nests and poisoning eggs. Since bluegills have a tendency to over-populate themselves, one method of control involves destroying the spawning beds in early summer. At the end of summer, all newly hatched fish can be eliminated by treating the marginal area of a lake in September during the middle of the day with rotenone. Tiny bream will be in this area to escape predation while larger, more desirable sized fish will be in deeper water at this time of day and not be affected.

III. FISH MANAGEMENT

A. Purpose - The purpose of fish management is to produce maximum yields of game fish of a desirable size in the shortest length of time.

1. Sports - This is the element of the population that purchases fishing licenses thereby funding the Game and Fresh Water Fish Commission. It is for this group that fishery biologists are most concerned with satisfying by producing the maximum yields as mentioned earlier.

2. Commercial - If fish supplies are not adequate for both kinds of fishing, then the sports fishery must have first consideration. Commercial fishing is desirable in a watershed if it does not intefere with sports fishing. The removal of undesirable or rough fish has no effect upon the game fish population. In the case of catfish, few of these are taken by sportsmen on hook and line. The problem of sports vs. commercial fishing usually comes to a head when the use of traps and baskets are allowed. Sportsmen believe that large amounts of game fish are taken illegally by these methods. Trotlines interfere with the operation of outboard motors and trolling for game fish. In the final analysis, if a controversy arises between these two types of fishing, commercial fishing must give way to angling.

3. Other recreation - Under the concept of multiple use, the fishery biologist must realize that there are uses of a lake and

its watershed other than for fish production and fishing. Considerations must be given to water skiing, boating, camping, swimming, hunting, and scuba diving.

B. Techniques

  1. Physical tests

    a. Mapping

    b. Temperature (stratification?)

    c. Turbidity

  2. Chemical tests

    a. pH

    b. General analysis

  3. Biological tests

    a. Fish population sampling

    b. Inventory of aquatic plants

    c. Types of invertebrates

    d. Extent of algal production

C. Equipment

  1. Shocker

  2. Minnow sein

  3. Plankton net

  4. Gill net

  5. Trammel net

  6. Haul seine

7. Water sampler
8. Bottom sampler
9. Slurp gun

D. Duties of Fisheries Biologist

1. Lake and stream surveys
2. Make field checks and keep records on growth rates, productivity, water chemistry, aquatic animals and aquatic plants.
3. Introduction of new fish species and maintaining proper balance between forage and predator species.
4. Investigation of parasites and disease.
5. Rehabilitate waters showing an overabundance of rough fish or a poor growth rate due to competition among the existing fish populations.
6. Collect and analyze creel census
7. Stocking recommendations
8. Recommendations pertaining to fishing laws.
9. Opening new fishing waters.
10. Keep the public informed of fisheries activities.
11. Cooperate with other agencies.

INVESTIGATION: THE STUDY OF AN ARTIFICIAL POND

PURPOSE: To increase the knowledge of the wide variety of living organisms found in this ecosystem, and to study some of their interrlationships. A secondary purpose is to develop some procedures and techniques of this type of investigation.

MATERIALS:

Plankton net
Spinning line
Vials
Squeeze bottles
Dip net (long handle)

PROCEDURE: PART I

1. Sample minute aquatic organisms by tossing a plankton net attached to a line into the pond and retrieve it.

2. The plankton will be concentrated in the bottle or test tube in the end of the net. Sample vials by washing with a squeeze bottle. Save for laboratory study.

3. Sample larger organisms with the long-handled dip net.

4. Sample organisms in and on the bottom by digging among weeds with the apron net.

5. Sample plants at and close to the water's edge, place between layers of newspaper.

6. Number all samples. Record the date, place, and environment of each.

7. Stake out an area of the pond to be surveyed and studied.

8. Make a wire loop 2 cm in diameter.

9. Drive a stake at any selected point within the area selected for study.

10. Stretch a 33.5 m tape.

11. Drop the loop at 30 cm intervals along the tape. Record any species found in the loop.

12. Run several lines and average the percentages.

MATERIALS: PART II & III

Secchi disc
Refractometer
Water sampler
Newspaper
Tape measure
Tags
Pencil
Wire loop
Stakes
Thermometer
Compound microscope
Classification book
Beaker
Petri dishes
Phenolphthalein indicator
Sulfuric acid ION
Methyl orange indicator
44 M sodium hydroxide

PROCEDURE: PART II

1. Take temperature reading of the water at different depths.

2. Record air temperature.

3. Use a Secchi disc to measure turbidity. Lower the disc into the water by a cord from the shaded side of the boat. Note and record the depth at which the white quadrants disappear. Lower the disc farther, then slowly raise until white quadrants reappear. Note this depth and average it with the first depth.

4. Lower the sampler to the desired depth and pull the stopper.

Allow the bottle to fill, save for tests in lab.

5. Estimate pH with pH paper.

Back in the Lab:

PROCEDURE II:

6. Examine plankton under a compound microscope.

7. Use a few drops of the plankton solution to inoculate a culture that you can later examine.

8. Study organisms preserved in the formalin vials.

9. Flatten and dry plants already placed in newspaper by placing them under a stack of books. Key out these materials later.

PROCEDURE: PART III

1. To determine alkalinity, measure 50 ml of the water sample into a beaker.

2. Add 5 drops of phenolphthalein indicator.

3. If the water turns pink, add N/10 sulphuric acid from a burette, drop by drop, stirring until the pink disappears. This is the end point. Note the burette reading. The number of ml used x 100 expresses the P or phenolphthalein alkalinity ($CaCO_3$) in parts per million.

4. Add 3 drops of methyl orange indicator and continue to titrate. Note the change if from yellow to pink.

5. When the end point is reached, again read the burette. The number of ml used x 100 will give the M alkalinity. If at the start no pink appears (when phenolpthalein is added), merely add the methyl orange

and titrate.

6. To determine CO_2 concentration, measure 100 ml of the sample into a beaker.

7. Add 5 drops of phenolphthalein indicator.

8. Titrate with N/44 NaOH until it turns pink. The amount of NaOH in ml used x 10 expresses the concentration of CO_2 in ppm.

STUDYING THE DATA AND CONCLUSIONS:

1. Where did the life, both animal and plant, in the pond probably come from?

2. What relation does the available supply in the pond have with the type of animal life present?

3. How does the percentage of oxygen and carbon dioxide affect the life in the pond?

SPECTRONIC 20 (SPEC 20) ANALYSIS OF WATER

Preparation of Standard Curve for Nitrate

The B. & L. Spectronic 20 offers the student of ecology a more reliable method of water analysis than the Hach Kit. The Hach chemicals can be used with the Spectronic 20 by referring to the handbook, Colorimetric Procedures and Chemicals for Water and Waste-water Analysis." Although the standard curves can be found in this text, a good student might want to prepare his own standard curve for nitrates, phosphates, and dissolved oxygen. This is a valid effort considering the variation or lack of consistency between one Spectronic 20 and another. In effect, the student would be "zeroing in" with the instrument he is using.

The following procedure is for the preparation of a standard curve for nitrate. The same general procedure can be used for phosphates and dissolved oxygen.

Step #1.

Calculation of per cent nitrate of standard
Potassium nitrate (KNO_3) is used here as the standard.
KNO_3

K = 39 (molecular weight)
N = 14 (molecular weight)
O = 16 x 3 48

101 total molecular weight

$$\frac{14}{101} = 13.9\%$$

This procedure yields the total nitrogen in KNO_3 as 13. 9%.

Since the Hach Kit Model N1-10 has a range of 1-10 ppm, it is necessary to prepare a curve within that range; hence the following formula:

$$\frac{13.9\%\ \text{nitrogen}}{100\ \text{mg}\ KNO_3} = \frac{100\%\ \text{nitrogen}}{X}$$

"X" is the number of milligrams of KNO_3 needed for a 100 ppm concentration of nitrogen. (one ppm = 1 mg/liter)

In this case X = 719 mg or .719 grams of KNO_3 to yield a 100 ppm reading in 1000 ml of distilled water.

Step #2.

Follow the procedure outlined on page 55-56 of the Hach calibrations referred to above. Before completing Step #2 be familar with setting up the Spec 20 as outlined below:

Zeroing the Spec 20:

1. Turn power on. Allow 5-10 minutes for warming up.

2. Select the wave length desired by turning the wave length control. (In this case 525 mm)

3. Adjust amplifier control with sample cover closed until needle reads 0 on the transmittance scale.

4. Place test tube sample of substance to be tested (without color) into the sample holder. Rotate the light control knob until the meter reads 100% transmittance. Repeat steps 3 & 4 until the same results are acquired at least twice in succession. The instrument is now zeroed and ready for reading of the prepared sample. It is important

that the instrument be checked frequently during the course of several sample readings. This is done by following the above procedure.

Step #3.

After mixing the sample with the Nitra Ver IV pillow, place a sample into the chamber for a reading. All reliable readings must fall between the 30% and 70% transmission numbers on the scale. This means, in most cases, it will be necessary to dilute the sample.

Dilutions can be made as follows:

$\frac{3}{4}$ sample	$\frac{1}{2}$ sample	$\frac{1}{4}$ sample
$\frac{1}{4}$ distilled H_2O	$\frac{1}{2}$ distilled H_2O	$\frac{3}{4}$ distilled H_2O

One of these readings will come close to the 30% transmission necessary. From that point a series of smaller dilutions should be made in order to obtain 4-5 readings within the 30-70% range. From these readings a graph can be prepared as a standard curve.

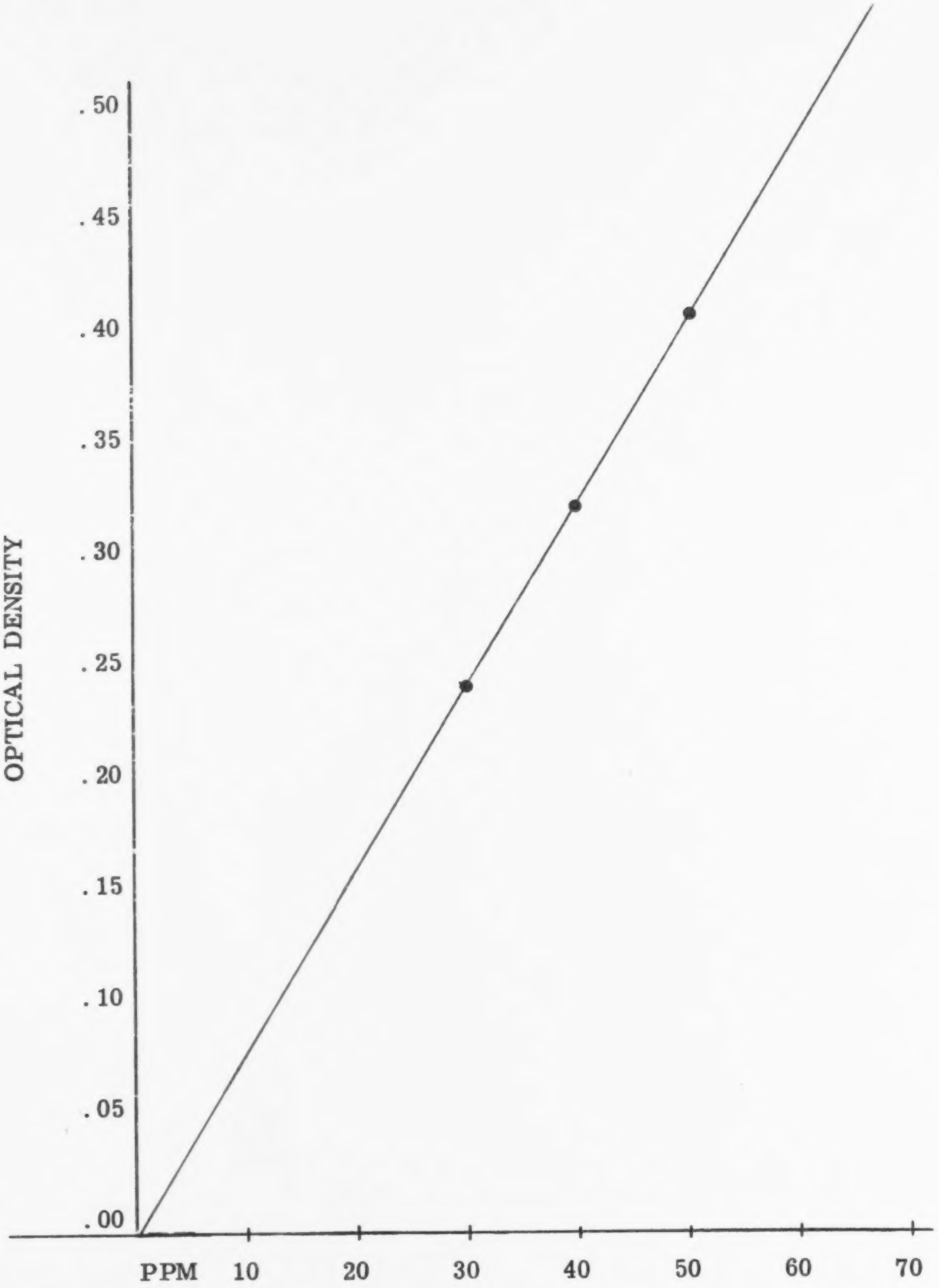
Example:

	100 ppm	- 1.
	75 ppm	- .62
In Range	56.25 ppm	- .42
	42.19 ppm	- .31
	31.64 ppm	- .225

Optical Density

Suggestion:

Prepare samples to permit duplicate points to be plotted on the graph. Thus, a double check on accuracy.



STANDARD CURVE FOR NITRATES

MAN VS. NATURE

BREVARD COUNTY ECOLOGY

The Indian River has no source nor mouth and is in fact not a river. Its geographical northern boundary starts approximately four miles north of Haulover Canal. The Haulover Canal is a man-made cut connecting Mosquito Lagoon with the body of water known as the Indian River. The Indian River serves as part of the Intercoastal Waterway from the Haulover Canal south to the St. Lucie Inlet. The Indian River is approximately two miles wide at its widest point. The length of the river is approximately one hundred miles. In addition to serving as the Intracoastal Waterway it is a recreational fishing area as well as an important commercial supplier of seafood. The sport fishing consists principally of the following fish, most of which are edible: trout, sheephead, drum, channel bass, and whiting. The Indian River is famous for the trout or spotted weakfish. Although most catches are under two pounds, more trout in the five pound category have been caught in the Indian River than any other place. This is because the area's water and temperature is most suited to the breeding and development of trout.

The commercial fishing consists of all the sport fish, plus mullet. The most important contributions from the Indian River to the seafood industry are clams, oysters, and blue crabs. The clams and oysters are harvested from shallow areas around Sebastian and also south of New Smyrna. The oysters are sold locally to restaurants and fishmarkets for retail distribution. The crabs are found in all areas of the river and are taken from crab traps. The blue crabs are sold live locally, and some are processed by local canners and shipped throughout the eastern United States and to New York's finest restaurants.

Starting approximately twenty years ago Brevard County's population began to grow. This growth developed along the Indian River because of the space industry build-up. As the population increased, the Indian River became polluted from sewage effluents, insecticides and fertilizer run-offs. Then the demand for more land came and builders filled in land from the river and the silt polluted the water even more and destroyed the breeding grounds of the fish. Then the seafood industry was hurt badly; the fish had few breeding grounds and the oysters were contaminated by the Escherichia coli, a bacteria causing destruction of oysters. The presence of this bacteria is determined by taking a 100 cc sample of water and it has more than 70 colonies of Escherichia coli, the shellfish are allowed to be marketed. Because this harmless bacteria flourishes in the human intestine, it is a good indicator of human waste pollution in the water. Oysters from moderately polluted areas may be purified by chlorination process used in the U. S. and England. It consists of washing and disinfecting the outside surfaces of the shell with chlorinated sea water, then placing the oysters for self-purification in sea water containing no residual chlorine. It is also most expensive and cannot be done on a commercial basis. The crabs, on the other hand, are not harmed inasmuch as they feed on sewage and decaying animal life. The crab is an economical shellfish, second only to the shrimp. The crabs found here are the blue crab and the very rare stone crab.

SHELLFISH

The "conditionally approved" shellfish harvesting area located in the south end of the County was closed once during 1970 in September, due to excessive rainfall; the area was closed from September 30 to October 24, a time of little or no commercial harvesting of oysters. With this exception, both shellfish harvesting areas remained open throughout the year with continued monthly sampling to ensure that water quality complied with the set standards. An updated shoreline sanitary survey of both the north and south shellfish harvesting areas as required by the Florida Division of Health and the U. S. Public Health Service was completed. This shoreline survey, along with the continuous monthly water sampling program, enables both areas to remain open and approved for the harvesting and direct marketing of shellfish.

During 1970, in cooperation with the U. S. Department of Interior, Dauphin Island, Alabama, laboratories and the Florida Division of Health, special water and shellfish meat samples were collected from within and near shellfish harvesting areas for analysis of pesticides and heavy metals. Analysis of the waters from the Indian River showed no pesticide residual from the ten most commonly used chlorinated hydrocarbons. The analysis of the water and meat tissues indicated no mercury present, and levels of other common heavy metals measured were within recommended limits.

SOLID WASTE DISPOSAL

The Department's effort to improve operation of the landfills and trash dumps in the area has been moderately successful. The county has taken over the complete operation of the landfill in the south area and provides equipment on a need basis for maintaining a cleared area for dumping at the Merritt Island trash dump. Intentional burning at these sites has been eliminated and the landfill is being operated as such. County operation of the central and north area landfills hopefully will become a reality early next year. Additional land for the landfill operations in the north and south areas is now being sought. A new site and location will be required in the north area. The interim operation of sanitary landfills until completion of the county-wide solid waste disposal project is necessary.

The proposed county-wide solid waste disposal project, after many delays, is now proceeding in the development and preliminary engineering phase. Enabling legislation passed previously for this project was amended to ensure that the project will serve the entire county on a mandatory basis and further to assure that an opposing community could not halt the project after the funds are committed. Modern refuse disposal centers and appurtenances equipped with suitable environmental protection systems will solve a critical county-wide solid waste disposal problem. The recommendations of the consulting engineers and cost estimates for the solution of the county-wide solid waste disposal problem were to be presented to the County in summer of 1971.

An ordinance requiring mandatory garbage and rubbish collection in the unincorporated areas of the County was adopted by the Board of County Commissioners and went into effect in 1971. This ordinance should greatly reduce the dumping of garbage and rubbish along the roadsides, ditches and waterways of the County on both public and private property. The many problems created, and clean-up expenses incurred, by unauthorized dumps and dumping of garbage over the countryside can possibly be eliminated by this ordinance. Many problems in enforcement of this ordinance are expected to surface and must be overcome if the desired effect of a cleaner community is to be obtained.

WATER POLLUTION CONTROL

The monthly water sampling program for basic data on the St. Johns River was continued at selected stations during the year, in cooperation with the Orange County Pollution Control Department. A summary of water quality findings from this cooperative sampling program was presented in a joint effort report to the Florida Department of Air and Water Pollution Control at a St. Johns River public hearing held in Palatka.

The U.S. Geological Survey Team under contract with the Central and Southern Florida Flood Control District completed studies of the physical, chemical, and biological conditions of the major tributary systems to the upper St. Johns River. This department provided assistance in portions of the biological studies on this survey. A report on their findings is under preparation and will be published soon.

The water quality monitoring and surveillance program on the Indian and Banana Rivers continued with special attention to the shellfish harvesting areas. A program for establishing qualitative and quantitave preliminary production levels of plankton and brackish water marine plants from

26 representative sampling stations in the Indian and Banana Rivers was established. This biological growth data study will complement the survey of bottom-living organisms and, when correlated with chemical and physical water quality parameters, will provide a new additional insight toward the measurement and control of water pollution. The bacteriological results of the department's sampling program on the Indian and Banana Rivers and ocean beaches over a four-year period, 1967-1970 were summarized into a report for the Division of Health. During this approximate four year period 2300 bacteriological samples were collected and analyzed. The results indicated excellent bacteriological water quality for recreational uses throughout the County.

During 1970, construction of the Pineda Causeway project under the Department of Transportation was begun. In the initial phases of construction the high volume of dredged materials resulted in excessive siltation and turbidity run-off into the Indian River north and south of the project site. During the dredging phases of this project, as a result of water quality monitoring for turbidity by this department at several locations in the immediately affected area, the dredging was halted upon request for turbidity and siltation control on two occasions. As a result of this action, the Department of Transportation experimented with a hanging skirt baffle (diaper) along the toe area of fill construction in an attempt to keep the heavier silt confined and settled in the causeway right-of-way. This method was only partially successful along the west shore of the Indian River especially during periods of heavy wind currents but was more effective on the protected eastern shore of the project. This method of attempting to control siltation and turbidity is in the early stages of development and experimentation in the State. The same method used later in this county on Department of Transportation dredge and fill road construction projects in waters protected from the wind minimized and maintained the siltation in a small selected area. The use of this baffle was successful at the Sykes Creek dredge and fill bridge project and also at the S. R. 528-401 widening and cloverleaf project near Port Canaveral. These two projects were accomplished in protected water areas.

A public hearing by the Florida Department of Air and Water Pollution Control for establishing stream classifications for the St. Johns River was held in the fall of the year. This department with support of the City of Melbourne and Board of County Commissioners requested that the St. Johns River from the Lake Washington Dam south to State Road 60 in Indian River County be classified as Class I - Public Water Supply, the present use. The St. Johns River north of Lake Washington in Brevard County is classified as Class III - Recreation, Fish and Wildlife Propagation. The Class I - Water Supply request was granted at a later date.

A public hearing by the U. S. Corps of Engineers on the proposed Sanford-St. Johns-Indian River Canal was held in the fall of the year. This department, from previous stream studies and water quality data obtained from the area to be affected, recommended that the Board of County Commissioners oppose construction of the project at this time. The department also entered written opposition to the project. Final determination on the project by the Corps of Engineers has, at this writing, not been made.

AIR POLLUTION

The results from the second year of operation of five air pollution effects stations were tabulated and forwarded to the U.S. Public Health Service for their review, information and comments. The results from these five air effects stations will indicate trends and effects of air quality over the years. These static type stations provide air quality parameter data on corrosion, sulfation, dustfall, wind-blown particulates, rubber cracking and nylon deterioration. The results from the third year operation of these stations are now being summarized and tabulated.

The annual average results from completion of two years' operation of a hi-vol air sampler for collection of suspended particulates for the Florida Air Monitoring Network indicate that airborne particulates in this area are among the lowest in Florida.

An improvement in air quality in the areas of the south landfill and Merritt Island trash dump was experienced when the County assumed the responsibility for operation or maintenance of the facilities. No intentional burning of trash or garbage takes place at these disposal sites. The County assumed operation of the north and central disposal sites early in 1971 and no burning permitted, hence burning dump problems in the County will be alleviated.

The majority of air pollution complaints continues to arise from poor operation of supermarket incinerators. A few of these supermarket incinerators have been replaced by trash compactors and/or bailers during the year. Plans have been made to replace more of these incinerators in the future by installation of compactor-bailer type facilities. In the fall of the year, a consultation to designate an intrastate air quality control region for the Central Florida area of which Brevard is a part was conducted by the National Air Pollution Control Administration of the U.S. Public Health Service. This consultation was requested by the Florida Department of Air and Water Pollution Control under the U.S. Clean Air Act. This first step to become part of a six-county air quality control region was supported by this department. The next step is formal designation of the proposed region, then issuance of air quality criteria to control concentrations of air pollutants harmful to health and damaging to property.

SEWAGE AND INDUSTRIAL WASTE FACILITIES

The year 1970 marked a period of slow but steady progress in the area of water pollution abatement. The completion and start-up of the county-owned South Brevard Beaches sewage treatment facility highlighted the year. This two million gallon per day wastewater treatment facility eliminated some 3500 septic tanks in the South Beaches area. This facility contains approximately 12 acres of evaporation-percolation ponds and is now successfully operating with no positive discharge to the Indian River.

During the year, two sewage treatment plants were phased out, Shaco Utilities and Surfside Elementary School. Both of the small systems served by these plants were connected to larger and more efficient sewage systems.

This department nominated the City of Melbourne sewage treatment facility for an award as the best operated high rate trickling filter plant within its class and population category in the state. The Melbourne facility received the best operated and maintained plant award.

The Brevard County Health Department, in cooperation with the Division of Health and Region Three, Florida Water and Pollution Control Operators Association, sponsored the Fifth Annual Short School, September 9 through 16, in the Health Department auditorium in Rockledge. As a result of the Division of Health's new compulsory certification requirement covering water and wastewater plant operators, this was the largest school yet. One hundred forty-one persons registered and the average attendance was 125 persons per session. The school's objective is to improve the quality of management, operation, and maintenance of all public and privately owned water and waste-water treatment facilities.

A total of 38 area water and sewage treatment plant operators participated in the semi-annual statewide Class "C" examination conducted by the department. Twenty-four operators successfully completed the examination and were awarded certificates by the Division of Health.

The lift station survey and inspection for all sewage systems was continued during 1970 with an additional 13 systems inspected. Deficiencies discovered during these inspections were brought to the owner's attention for correction; the result has been a decline in the number of lift station failures and subsequent sewer system overflows in the County.

During 1970, this department, in cooperation with, and based upon, recommendations from the Division of Health and the Department of Air chromium compounds in cooling towers in the County. Elimination of chromium compounds and use of less objectionable compounds is possible. Surveillance was continued to ensure that chromium compounds were not being used at facilities previously requested by this department to discontinue their use.

During the latter part of the year, in cooperation with the Department of Air and Water Pollution Control, the department distributed application forms for operating permits to all sewage treatment plants in the County. Operation permits for all sewage treatment facilities with a capacity of at least 3,000 gallons per day are now required by law.

Waste treatment plant expansions or additions for 1970 included:

1) Central Section
   - a. Cape Kennedy Air Force Station added a new 15,000 gallon-per-day package-type extended-aeration plant at its museum site.
   - b. Kennedy Space Center added a new 14,000 gallon-per-day extended-aeration package plant at its unified (S) Band location.
   - c. Shaco plant in West Cocoa was phased out. The sewage from the area previously served by this plant is now pumped to the City of Cocoa system.

2) South Section
   - a. Brevard County completed and put into operation the two

million gallon per day South Beaches contact stabilization plant. The facility includes approximately 12 acres of evaporation-percolation ponds. This plant has no positive discharge of final effluent; evaporation, percolation and irrigation will be utilized in an attempt to maintain a no-discharge status.

b. Gulf American Corporation at its Barefoot Bay Subdivision development placed into operation a temporary aeration package-type treatment plant and evaporation-percolation pond with no discharge.

Kenlock, Brevard County Health Dept., 1970.

SEWAGE TREATMENT

BACKGROUND: After sewage is collected in public sewers and brought to a central point, it may receive only primary treatment or perhaps primary and secondary treatment. In a few instances it may also receive tertiary treatment. These are general terms used to describe the degree to which waste water is cleaned before it is put into a river or lake or used again. Since sewage treatment plants are not all alike, you may see different methods of treatment than those mentioned here if you visit the treatment plant in your community.

If we continue to use water to move sewage and organic wastes and still expect relatively clean streams, rivers, and lakes; we must properly process our huge quantities of sewage and wastes so they will not pollute streams. This is the purpose of sewage-treatment plants. Cities and towns usually construct and operate their own central sewage-treatment plants. In addition to receiving the sewage from homes, hospitals, garages, hotels, and other businesses; they generally serve some industries. However, numerous industrial plants maintain their own facilities for treating sewage before redirecting the water they've used back into the river.

Although new plants and additions to existing plants are being constructed, cities and towns generally are not building sewage-treatment plants fast enough to keep up with the need for them. Many cities and towns use sewage-treatment plants designed and built years ago, and these are overloaded as the cities and towns grow in size and people use more water.

In many large cities, storm drains built to handle the runoff from city streets flow directly into the sewer system. When there is much rain, the great amount of runoff cannot be taken care of by the sewage-treatment plant, so some effluent flows directly into a river or lake, carrying raw sewage along with it. Ideally, storm-drainage systems and sewer systems should be completely separate, but this is a very expensive type of operation.

The newer city and urban type of design planning insists on separation of utility drainage and sewage systems but most planning studies have shown that the cost is too prohibitive to attempt redesign of a large city combined facility.

The decision to the specific method of treatment to be used, depends largely on the strength and quantity of the sewage in relation to the nature and volume of the water (river, stream, lake, reservoir) into which the treated waste water is to be discharged.

Primary Treatment. This mainly involves removal of the solids from waste water. This type of treatment is the only kind many towns use, but there are different methods of accomplishing it. The first step in primary treatment is usually some type of screen to trap the sticks, rags, and other large objects. Or all the sewage may pass through a grinder that chops up these large objects. In the next step, the sewage moves slowly through a grit chamber where stones, sand and other heavy inorganic materials sink to the bottom and then are removed from the chamber. Next, the waste water--also called effluent--goes to a settling tank; it stays there long enough for organic matter and fine particles of other material to settle so they can be collected, and to allow scum and grease to float to the surface where they are skimmed off. Certain chemicals can be added to the settling tank to cause the fine particles to cling together and settle out faster.

In primary treatment, the effluent from the settling tank is discharged into a river or stream or allowed to soak into the land. Sometimes, as the effluent flows out of the settling tank, it is treated with chlorine to kill harmful bacteria.

The collected solids--called sludge--from the bottom of the settling tank then go to a sludge chamber or digester where decomposing bacteria go to work on them. The digested sludge then goes to a drying bed and after it is dry it may be burned or buried or it can be put on the land as a soil conditioner-fertilizer.

In terms of reduction in Biological Oxygen Demand, Primary treatment results in a 40% reduction (approximate).

Secondary Treatment. Often, the effluent resulting from primary treatment is not clean enough, so secondary treatment must be practiced. In secondary treatment, the waste water goes through all the steps in primary treatment and then through one of two processes for further organic decomposition of wastes. Both processes depend upon biological action and both require oxygen, the oxygen is supplied by spraying the effluent into the air or by pumping air into it (aeration).

In one of the processes, the effluent goes from primary settling tank to a trickling filter in which it passes slowly over stones or other material where biological growth decompose the waste still in the effluent. The purpose of the stones and other material in the trickling filter is not to filter out the solids but to provide as much surface area as possible where there is oxygen so that the biological growths can live and do their work in the other basic secondary process, effluent from the primary settling tank goes into a sludge tank where activated sludge--material that has various biological growths in it--completes the process of decomposing organic materials. While the effluent remains in the sludge tank, it is continuously aerated.

The effluent from either the trickling filter or the activated sludge tank then goes to a secondary settling tank to the sludge chamber or digester. As it flows from the secondary settling tank, the effluent is treated with chlorine before being released into a stream, river, or lake, or being allowed to soak into the earth.

BOD is reduced an additional 45-55% for a total of 85-95% BOD removal. Costs mount rapidly when 90% BOD removal is approached.

Tertiary Treatment. But even secondary treatment doesn't get waste water clean enough in some situations. So tertiary treatment is used after the waste water goes through primary and secondary treatment. After tertiary treatment, the waste water is actually clean enough to be run through a city's water-treatment process for water to be used in homes.

Very little waste water now receives tertiary treatment, and there is no typical tertiary treatment plant. The process used depends upon the specific need for further treatment of the effluent after it has received secondary treatment. Tertiary treatment consists of slow or rapid filtering of the effluent through sand to remove dissolved solids. It could be aeration to foam out detergents. It might be by use of chemical precipitation with alum or silica to settle out solids. Or it could be superchlorination followed by dechlorination to ensure killing of harmful bacteria and disease-bearing organisms.

One important thing to remember is that waste water properly treated is no longer water wasted. It is good water and can be used again. Another important fact is that treatment of waste water helps prevent the great damage that sewage and organic wastes do when they get into streams, rivers, and lakes. As we traced the different methods of treating waste water, you may have observed that all sewage treatment is similar to nature's endless chemical and physical water-purifying processes. But nature's processes take a long time and they simply cannot take care of the huge amounts of waste man wants to get rid of each day. Primary, secondary, and tertiary waste-water treatment does the same thing nature does, only faster and under controlled conditions. Why don't all cities and industries treat their waste water so they can use it again? It is primarily a matter of high costs.

Soil and Water Conservation, Boy Scouts of America, New Jersey, 1968.

INVESTIGATION: USE OF THE RINGELMANN CHARTS

OBJECTIVE: To determine the density of smoke by the use of Ringelmann charts.

BACKGROUND: In the burning of any material if complete combustion takes place the by-products are carbon dioxide and water vapor. The products are colorless and harmless. In actuality, it is usually impossible to burn any product without some other by-products than CO_2 and water vapor even if excessive oxygen is supplied. Thus the unburned carbon particles combine with other gases to produce various shades of gray.

To gauge the amount of pollution being discharged a Power's Microringlemann or a Ringlemann chart can be used in a series of one through five.

PROCEDURE:

1. Select site to be measured. Stand more than 100 feet from site. Make sure the stack or site background is clear of buildings.

2. Hold the Ringelmann's Chart by the right hand corner between thumb and index finger with reproduced grids toward the stack.

3. Face the stack and hold it at arm's length. Move the chart back and forth until the smoke matches the shade of one of the pairs of grids.

4. Observe over a period of time using incruments of 1/2 minute for 15 minutes.

5. Record number of observations and each Ringelmann number.

NUMBER OF OBSERVATIONS	(TIME) INTERVAL	RINGELMANN'S NUMBER	TOTAL
1.			
2.			
3.			
4.			
5.			

TOTAL PRODUCT = ____________

6. Multiply number of observations by the chart number. Total the products and divide by the number of observations. This will give you the smoke density.

$$\frac{\text{TOTAL PRODUCT}}{\text{TOTAL OBSERVATION}} = \frac{\text{SMOKE}}{\text{DENSITY}}$$

Example:

Ringelmann #		Observations	Total
1	X	5	5
2	X	7	14
3	X	8	24
4	X	10	40
		30	83

83/30 = 2.7 ≃ 3 (smoke density)

MAN VS. NATURE

1. How would you feel about living in a circular condominium or a circular high rise? Where?
2. How many people are presently living at your home?
3. What are the ages of your children?
4. Would you be in favor of one High School, one Junior High School, and one Elementary School? If so, where would you locate them?
5. What, in your opinion is a fair price to pay for the condominium?
6. What do you think of three large motels instead of the scattered ones we now have in town?
7. Are you in favor of the mass transit program, such as the monorails? Why or why not?
8. How many cars do you now presently have? If more than one would you consider giving them up for one car per family?
9. Would you be in favor of a centralized shopping center? If so, where?
10. Would you consider buying or renting a condominium?
11. How and where would you recommend the recreational areas to be built?
12. How many rooms do you have in your home?

Add any other information that you think is necessary to this questionnaire.

INVESTIGATION: PARTICLE CONTAMINATION OF AIR

PURPOSE: To show particle contamination of air.

MATERIALS:

- Several 1 gallon glass or plastic jars from school cafeteria
- Balance
- Wash bottle
- Evaporating dish
- Distilled water

PROCEDURE:

1. Wash jars carefully--rinse with distilled water. All detergent must be rinsed out. Rinse well.
2. Put approximately 4" of distilled water in each jar.
3. Locate jars in open area, above ground level by several feet (perhaps the roof of the school) to stop material from being blown in from the surface.
4. Leave jars in place for 30 days. Add distilled water from time to time. Do not allow the jars to dry out.
5. (a) Collect water--wash jars with distilled water. Add this to water taken from jar.
   (b) Weigh evaporating dish--evaporate water slowly.
   (c) Weigh dish--subtract first weight from second.
6. Compare results from different areas with respect to:
   (a) Temperature
   (b) Wind direction
   (c) Industrial locations
   (d) Rainfall

(e) Fires

(f) Hurricanes

7. Compute the area of the jar mouth--convert to square meters. Calculate the amount of particles that fall each month, year, decade.

INVESTIGATION: DETECTING OF ATMOSPHERIC CARBON MONOXIDE

BACKGROUND: It is difficult to detect carbon monoxide and other odorless and colorless gases in a simple manner. There are various methods for detecting carbon monoxide. One method is used by the National Bureau of Standards Colorimetric Indicating Gel. The Gel is composed of silica gel permeated with ammonium molybdate, sulfuric acid and palladium chloride. The yellow silico-molybdate complex is formed, and the palladium serves as a catalyst that reduces to carbon monoxide. One can tell that carbon monoxide is present if the gel turns from yellow to green or blue. The gel will turn either blue or green depending on the amount of carbon monoxide present. Silica Gel Refrigeration (6-12 mesh) from Central Scientific Company.

PURPOSE: To be able to detect carbon monoxide in the air.

MATERIALS:

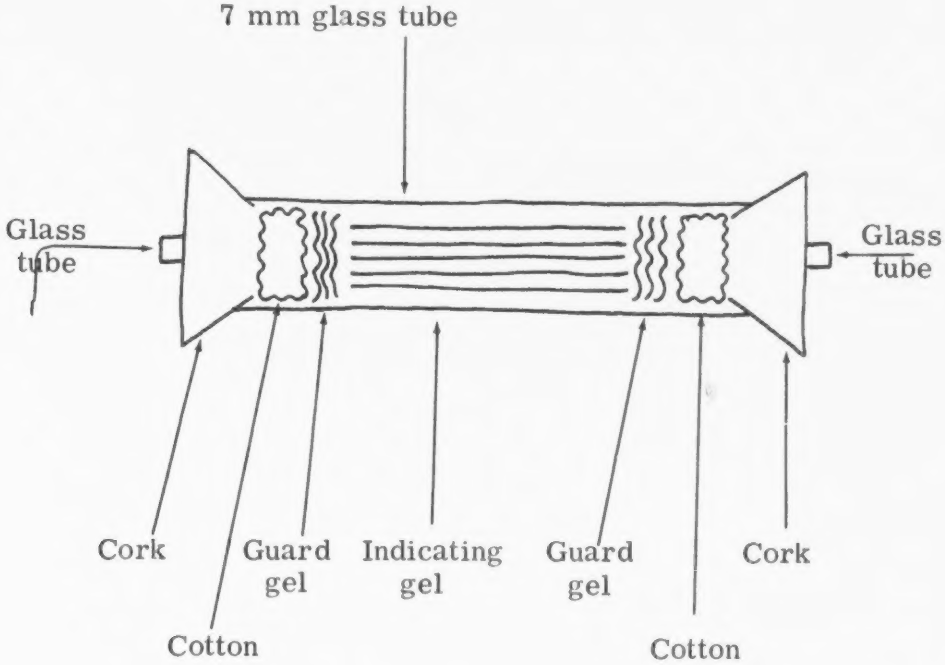
Vacum pump
Absorbent cotton
Guard Gel
Silica Indicating Gel
7 mm glass tube
Cork
7 mm glass filter tube
Indicator gels #38530
Size 1P - Silica Gel indicating (6-16 mesh) #38532

PROCEDURE:

1. Clean all glass to be used with sulfuric acid and rinsing with distilled water.

2. Place a small wad of absorbent cotton into one end of the glass tube and insert cork.

3. Fill the tube with 5 cm length of guard gel.
4. Add 2 cm of indicating gel.
5. Add a second cm length of guard gel.
6. Insert cotton wad and cork.
7. Insert glass tube into cork.


7 mm glass tube
Glass tube
Glass tube
Cork
Cotton
Guard gel
Indicating gel
Guard gel
Cotton
Cork

8. Connect one end of glass tube to vacum by a hose.

9. Pump air through tubing and gel. Note color change. A color change to blue or green indicates the presence of carbon monoxide.

Bell, F. A. Jr., N. B. S. Detector Tube Method for Carbon Monoxide in Air, Technical Assistance Branch, Division of Air Pollution, R. A. Taft Sanitary Engineering Center, 1961.

INVESTIGATION: DETERGENTS

BACKGROUND: Detergents are complex organic chemicals that are often used as a substitute for soap. If household waste water is properly processed during sewage treatment or in a septic system, the effluent will contain only limited amounts of detergent. However, since most detergents break down to release varying amounts of phosphate, even treated water poses an environmental hazard. This is true because a lack of phosphate is usually the limiting factor in inhibiting excessive algae growth.* Phosphate regulation of natural waters is easier to obtain than the limitation of nitrogen or carbon, due to the large atmospheric reserves of these two elements in the form of CO_2 and N_2.

"Improved" detergents have been developed. These detergents have less phosphate or none at all; however, the cleansing properties of detergents seem to be closely allied with higher phosphate content. The detergents which lack phosphate, contain excessive nitrogen which can be harmful to certain natural waters. Another property of a desirable detergent is biodegradability. This means that the detergent molecule is very susceptible to the action of bacterial enzymes and will therefore break down during water treatment. If detergents are not degraded they may form an unsightly foam in rivers or lakes and their presence there could be directly toxic to fish and other aquatic life.

PURPOSE: To detect the presence of detergents at water study sites.

MATERIALS: Use the Model DE - 2 detergent test kit sold by the Hach Chemical Company.

PROCEDURE:

1. Fill one of the test tubes to the upper mark with the water to be tested.

2. Add 12 drops of Detergent Test Solution and shake to mix.

3. Add chloroform to the lower mark on the test tube. (Chloroform is heavier than water and will sink.) Stopper and shake vigorously for 30 seconds and allow to stand for 1 minute to allow the chloroform to separate.

4. Using the draw-off pipette, remove the water from the tube and discard.

5. Refill the test tube to the upper mark with the Wash Water Buffer and discard. This step washes away the remaining water sample.

6. Refill the test tube to the upper mark with the Wash Water Buffer, stopper and shake vigorously for 30 seconds. Allow to stand for 1 minute to allow the chloroform to separate.

7. Insert the test tube containing the prepared sample in the opening nearest the middle of the color comparator.

8. Fill the other test tube with demineralized water and place it in the other opening in the comparator.

9. Hold the color comparator up to a light, such as the sky, a window or a lamp and view through the two openings in the front. Rotate the Detergents Color Disc until a color match is obtained. Read the ppm detergents (LAS and/or ABS) from the scale window.

10. If the color is darker than the highest reading on the color disc,

the sample may be diluted 20 to 1 by adding 1/ml of sample to the test tube (using the plastic dropper, filled to the top, or 1 ml mark), and filling the test tube to the upper mark (20 ml) with demineralized water. Repeat steps 2 through 8 and multiply the results by 20.

NOTE: If the water sample is turbid, the chloroform layer must be filtered after step 6 using the procedure given below.

a. Place a small ball (about the size of a large pea) of glass wool in the filter thimble.

b. Using the draw-off pipette to transfer the chloroform into the extra test tube,

c. Proceed with step 7.

d. Enough Wash Water Buffer is included for 32 tests. Enough Detergent Test Solution and Chloroform are included for approximately 90 tests.

*It will be remembered that excessive algae growth severely limits the oxygen available to fish and other animal life, especially at night or during cloudy weather.

INVESTIGATION: BIOCHEMICAL OXYGEN DEMAND

BACKGROUND: A high Biochemical Oxygen Demand (BOD) indicates that a great amount of oxygen is needed for bacteria and mold for the decomposition of a large amount of dissolved organic material. If the BOD is very high, the available amount of dissolved oxygen is utilized for decomposition and there is little left for larger animal and plant life. A BOD test can be affected by temperature, agitation, etc., but it gives a good estimate of the amount of decomposing activity that a body of water must support.

To perform the test collect samples in glass pint jars obtained from home or in 250 ml glass-stoppered bottles from the laboratory. However, all containers should be clean and similar in size. Locate collection points on a stream or lake. At each point rinse a collecting bottle several times in the water to be sampled. Then fill the jar to overflowing, cap it or stop it under water so that when the bottle is tipped, no free air bubbles can be seen.

Another procedure that may be necessary is to filter those samples which have visible algae or crustacean life within. Since BOD is essentially a measure of microscopic bacterial decomposition process, larger organisms in some samples may affect testing conditions and results. Thus, filter the sample through paper toweling or cloth as it is collected to remove the visible organism.

At the same time samples are collected, you should test for dissolved oxygen at the same sample site and record the results. Use the

Hach DO test kit listed below.

The closed bottles should be identified according to test site designation and allowed to sit undisturbed in the dark at constant temperature for five consecutive days.

During this period, bacteria in the water will use up oxygen in the process of decomposing organic material in the water. The amount of oxygen consumed is then a measure of the amount of organic material in the water.

At the end of five days a second dissolved oxygen test should be completed for each sample bottle, and compared with the initial results. Subtract the ppm of oxygen found in the second BOD test from the first test made at the collecting site.

The difference in amount of dissolved oxygen will be a measure of the BOD and will indicate the amount of organic decomposition occurring in the water.

PURPOSE: To determine the quality of natural waters with respect to its O_2 requirement ofr organic decomposition.

MATERIALS: Those materials contained in Hach Kit Model OX-2P

PROCEDURE:

A. High Range (1 drop = ppm DO)

1. Fill the glass stoppered DO bottle with the water to be treated by allowing the water to overflow the bottle for 2 or 3 minutes. Be certain there are no air bubbles present in the bottle.

2. Add the contents of one pillow each of dissolved Oxygen 1 Powder

(Manganous Sulfate) and Dissolved Oxygen 11 Powder (Alkaline Iodide-Azide). Stopper the bottle carefully sothat air is not trapped in the bottle. See Note A. Grip the bottle and stopper firmly and shake vigorously to mix. See Note B. A flocculant precipitate will be formed. If oxygen is present the precipitate will be brownish orange in color.

3. Allow the sample to stand until the floc has settled halfway, see Note E, and leaves the upper half of the bottle clear. Then again shake the bottle and again let it stand until the upper half of the bottle is clear.

4. Remove the stopper and add the contents of one pillow of Dissolved Oxygen 111 Powder (dry acid). Carefully re-stopper and shake to mix. The floc will dissolve and a yellow color will develop if oxygen was present. This is the prepared sample.

5. Fill the plastic measuring tube level full with prepared sample and pour it into the mixing bottle.

6. While swirling the sample to mix, add PAO dropwise, counting each drop, until the sample changes from yellow to colorless. The dropper must be held in a vertical manner. Each drop is equal to 1 ppm Dissolved Oxygen.

7. Repeat this procedure on the same sample after having stored in the dark for 5 days.

8. BOD = first O_2 ppm minus 2nd O_2 ppm

B. Low Range (1 drop = 0.2 ppm DO)

If the result from step 6 is very low, such as 3 ppm or less, it is advisable to test a larger sample so as to obtain a more sensitive test.

This may be done by titrating directly in the DO sample bottle as follows:

7. Using the prepared sample left over from step 4 above, pour off the contents of the DO bottle until the level just reaches the mark on the bottle.

8. PAO dropwise, counting each drop, until the sample changes from yellow to colorless. Each drop of PAO added is equal to 0.2 ppm Dissolved Oxygen in the sample.

NOTES:

A. It is a bit tricky to stopper the DO bottle without getting an air bubble trapped in the bottle. To avoid the air bubble, incline the DO bottle slightly, and insert the stopper with a quick thrust. This will force air bubbles out. If air bubbles are trapped in the DO bottle in steps 2 or 4, the sample should be discarded and the test started over.

B. A small amount of powdered reagent may remain stuck to the bottom of the DO bottle at this point, but this will not affect the test.

C. Do not allow the PAO solution to stand in direct sunlight, as it is decomposed by ultraviolet radiation.

D. If DO is to be determined in sewage, pretreatment with Copper Sulfate-Sulfamic Acid is required. Write for instructions. The following items are necessary for this treatment:

1949-00 Cylinder, graduated, 500 ml -- each 7.95
357-13 Copper Sulfate-Sulfamic Acid -- 4 oz DB 1.40
1864-99 Siphon -- each 2.00

Above items also come in a DO in Sewage Test Kit, Model OX-13, Cat. No. 2380-00, each $38.95.

E. In samples that contain high concentrations of chloride such as seawater, this floc will not settle. However, no interference is observed as long as the sample is allowed to stand in contact with the floc for 4 or 5 minutes.

OPTIONAL: Methylene Blue is an indicator of the BOD level in water. It turns pale blue to white or clear as a result of a lack of oxygen for cellular respiration. The speed with which a sample changes color indicates the relative BOD level in the water.

Samples can be collected in pint jars in a manner similar to the five-day BOD test. Five separate stoppered or capped test tubes should be prepared from each different sample. Label each according to the collection site. A very dilute solution of methylene blue test solution should be prepared--20-25 drops of commercial solution per 1/2 pint or 250 ml of water to make a stock solution. Place 1, 4, 8, 12 and 16 drops of stock solution in some tube each of the set of five prepared. Labeling should also include the number of drops of methylene blue in each tube.

Students should check the test tubes each day at the same time for clearing. Frequently, it will be helpful to place the tubes against a white sheet of paper to note the slight variations. The results can be charted on Data Sheets. The set of tubes that clears the fastest is that with the most need for oxygen and therefore has the highest BOD.

METHYLENE BLUE TEST
STUDENT DATA SHEET

Test Site	Drops of Methylene Blue Solution				
	1	4	8	12	16
1st day					
2nd day					
3rd day					
4th day					
5th day					

Name (s) ____

BASELINE DATA FOR WATER ANALYSIS*

I. COLIFORM BACTERIA

A. Oyster beds - These locations are closed to harvesting if any one of the following conditions exist:

a. Three or more inches of rain in any 72 hour period.

b. The media number of bacteria counted at - 21 stations (per bed) exceeds 70 colonies per 100 ml of water tested.

c. 10% of the stations exceed 230 colonies per 100 ml of water tested.

B. Sewage effluent - Must be maintained at less than 1000 coliform colonies per 100 ml of the water tested. (See item 4 - Detergents).

C. Water utilized for drinking and/or swimming - Should have less than one or none (no colonies on the test plate).

II. PHOSPHATES (Ortho)

- Mean values unknown

III. NITRATES & NITRITES

A. Mean values in the Indian River 5-6 ppm.

B. Mean value for sewage effluent, 10-15 ppm.

IV. DETERGENTS

Probably at a level of very much less than 1 ppm in the Indian River.

*All data obtained from the State Board of Health office at Rockledge, Fla.

Sewage effluent is presumably also low since our detergents are biodegradable.

V. BIOCHEMICAL OXYGEN DEMAND (BOD)

The mean level in the Indian River is about 2-3 ppm, the River is usually about 90% saturation with respect to oxygen. An unusually high value for a BOD would be 10-15 ppm. Readings are made from samples taken at a depth of 3 ft.

VI. ALGAE & PROTOZOA

Indian River
February, 1971

Number per ml

Green	Diat.	Dino.	Prot.	Date	Sta.
		(No data available)			9
					16
					22
					22A
					25
					30
					35
40	160	560	360	2/2	45
160	40	40	40	2/11	50
40	760	40	40	2/18	53
200	600	80	40	2/18	54
1200	160	400	80	2/4	55
120	200	150	40	2/11	56
80	400	40	80	2/4	57
40	120	40	40	2/4	58
					76
200	1100	320	160	2/15	81
80	400	200	280	2/22	98
40	120	40	320	2/22	104

Indian River
April, 1971

Number per ml

Green	Diat.	Dino.	Prot.	Date	Sta.
		(No data available)			9-16
280	5000	120	80	4/12	22
200	4200	120	80	4/12	22
40	440	40	40	4/12	25
40	320	40	40	4/12	30
40	1100	20	40	4/12	35
					45
280	80	40	40	4/12	50
40	520	40	80	4/9	53
80	440	40	40	4/9	54
240	240	160	40	4/12	55
160	3700	40	40	4/26	56
400	13000	240	80		57
					58
				4/22	76
				4/22	81
				4/23	98
120	600	80		4/23	104*

*Plankton

St. John's River

Station 8

Number per ml

Date	10/5	11/2	12/14	2/8	3/8	5/10	6/14
Green	40	40	600	80	560	320	44,000*
Blue g.	40	40	40	160	40	40	40
Diatoms	40	40	40	120	200	40	40
Protozoa	40	40	40	40	40	40	40
ppm PO							0.3
Coliform	460	N00 +		93	1100 +	460	

6/14 *Large flagellates

St. John's River

Station 38

Number per ml

Date	10/5	11/2	12/14	2/8	3/8	5/10	6/14
Green	2400	4800	4500	20,000	2400	5200	280
Blue g.	8800	8300	12,500	14,000	16,000	34,000	6800
Diatoms	2600	2900	8000	15,000	4600	2800	880
Protozoa	320	40	1300	3,000	160	40	40

INVESTIGATION: NITRATES AND PHOSPHATES (HACH METHODS)

BACKGROUND: Plants, fish, water, rocks, silt and solid wastes are all easily recognized in natural waters. Chemicals, however creep in unnoticed if they are colorless and odorless. The presence and source of chemicals can be detected by testing the water. Possible sources and effects of several chemicals found in water are listed in Table I.

Table I

Possible Sources and Effects of Several Chemicals Found in Water

Chemical	Possible sources	Effect of excess	Standard natural concentration at site
Ammonia Nitrogen	Decomposition of organic matter Fish urine	Increases bacterial growth Reduces growth of fish Increases oxygen consumption of fish	
Nitrate	Fertilizers Decomposition of organic matter Industrial acids	Increase growth of algae and higher plants Possible cause of "blue babies" in certain concentrations	
Phosphate	Fertilizers Household detergents Organic matter	Increases algal growth	

Phosphates and nitrates are normal components of all natural waters. Moderate quantities of these compounds are required for the normal growth of aquatic plants, terrestrial plants, and algae. Excessive amounts of these chemicals are often found in polluted natural waters resulting in the hypernutrition (eutrophication) of the algae or aquatic plants. This overfeeding, or fertilization, usually results in increased plant growth, an algae "bloom" or a weed choked body of water. Since most organisms utilize oxygen for respiration, the overgrowth of weeds or algae will compete with fish for oxygen often resulting in a massive fish kill. Eutrophication often results in the "death" of the body of water filling it with dead aquatic plants and killing its aquatic animal life. See the text and study the nitrogen cycle for a thorough understanding of the role of nitrogen in the ecosystem.

Bacteria can convert nitrogen as follows:

Organic Nitrogen → decay → Ammonia → Nitrite bacteria → Nitrite → Nitrate bacteria → Nitrate

PURPOSE: To learn how to determine the amount of nitrate, nitrite, and ortho phosphate in water for the purpose of making such tests on samples collected at school or home study sites.

MATERIALS:

Hach kit Model NI-10 (Nitrate-Nitrite Test Kit)
Hach kit Model PO-19 (Phosphate Test Kit)

PROCEDURE: Total Nitrate-Nitrite (does not include ammonium or organic nitrogen).

PART I: Nitrate - Nitrite

1. Fill one of the color viewing tubes about halfway to the lower mark with demineralized water. Stopper and shake vigorously. Empty the tube and repeat the procedure.

2. Fill the pipette by suction to just above the constriction, with the water sample. The tip of the pipette is then wiped clean and the excess liquid alowed to drain, automatically stopping at the constriction. For best results, rinse the pipette several times with the sample. Blow to discharge the sample from the pipette into the rinsed color viewing tube.

3. Fill the color viewing tube to the upper mark (10 ml) with demineralized water.

4. Add the contents of one Vitra Ver IV Powder Pillow, stopper the tube and shake vigourously for one minute. If nitrate and/or nitrite is present, a pink color will develop. Allow an additional 3 minutes for full color development.

5. Insert the tube containing the prepared sample in the right hand opening on top of the color comparator.

6. Fill the second color viewing tube to the lower mark with demineralized water and insert it in the left hand opening of the color comparator.

7. Hold the color comparator up to a light, such as the sky, a

window, or a lamp and view through the openings in front. Rotate the color disc until a color match is obtained. Read the ppm Nitrate Nitrogen (N) and/or Nitrite Nitrogen (N) from the scale window. See Notes 1 and 3.

Medium Range (0-10 ppm Nitrogen)

1. Same as step 1 above.

2. Rinse the plastic dropper with the sample or with the pretreated sample, then fill to the 1.0 ml mark. Add it to the rinsed color viewing tube.

3-7. Same as steps 3 through 7 above, except that the scale reading is divided by ten to obtain the ppm Nitrate and/or Nitrite Nitrogen (N) in the sample.

Low Range (0-1 ppm Nitrogen)

1. Rinse a clean color viewing tube with some of the water to be tested, then fill it to the upper mark with the water sample. No dilution is required.

2-5. Same as steps 4 through 7 above, except that some original water sample should be used instead of demineralized water in step 6 if there is color and/or turbidity in the water itself and in step 7, the scale reading is divided by one hundred to obtain the ppm Nitrate and/or Nitrite Nitrogen (N) in the sample.

PART II: Phosphate

High Range (1-50 ppm Orthophosphate)

1. Rinse the plastic dropper several times with the water sample.

2. Fill the dropper to the 0.5 ml mark. Discharge into one of the color viewing tubes, which has been rinsed with demineralized water.

3. Add demineralized water to the 5 ml mark. Swirl to mix.

4. Add the contents of one Phos Ver 111 Powder Pillow for 5 ml sample. Swirl to mix. Allow one minute for color development. If phosphate is present, a blue-violet color will develop.

5. Insert the tube of prepared sample in the right opening on top of the color comparator.

6. Fill the other tube to the 5 ml mark with demineralized water. Insert it in the left opening of the color comparator.

7. Hold the color comparator up to a light such as the sky, a window or a lamp, and view through the two openings in the front. Rotate the color disc until a color match is obtained. Read the ppm phosphate (PO_4) from the scale window.

Low Range (0-5 ppm Orthophosphate)

1. Fill both color viewing tubes to the 5 ml mark with the water sample.

2. To one of the tubes, add the contents of one Phos Ver 111 Powder Pillow for 5 ml sample, and swirl to mix. Allow one minute for color development. If phosphate is present, a blue-violet color will develop.

3. Insert the tube of untreated water sample in the left opening of the color comparator.

4. Hold the color comparator up to a light such as the sky, a window, or a lamp and view through the two openings in the front. Rotate the color disc until a color match is obtained. Divide the reading in the scale window by 10 to obtain the ppm Phosphate (PO_4).

NOTES:

A. The color should be compared after one minute but before two minutes.

B. To obtain the value as ppm Phosphorus (P), divide the Phosphate (PO_4) value by 3.

Reference: Hach Chemical Company
Box 907
Ames, Iowa 50010

BREVARD COUNTY PHOSPHATE
CONCENTRATION DATA

Considering that Concentration Data does not exist for phosphate levels in various levels in various streams or bodies of water in the county, it is an excellent opportunity to obtain meaningful data from all of the schools as the Ecology Course develops and significant testing is undertaken. To encourage this accumulation of data, it is requested that each teacher commence logging of data in the following table at the earliest possible opportunity. The data will be reviewed at an early in-service teacher meeting.

Date	Time	River, Stream, or Lake	Detailed Location	No. of Tests	Ave. Phos. (PPM)	Comments on Other Conditions

INVESTIGATION: A STUDY OF THE SOURCES OF WATER POLLUTANTS

BACKGROUND: A river drainage canal, or lake receives its supply of water from many sources. One of these is surface runoff as a result of rainfall. This runoff might originate in a field, forest, lawn, grove, parking lot, or even on a highway. In towns or cities the runoff is often collected in storm sewers which conduct this effluent by way of underground pipes to some natural body of water for disposal. The effluent pipes of sewage disposal plants, or the drain fields of septic systems, also make additions to the runoff received by our lakes, ponds, streams, and canals. The pollutants entering these waters must orginate from runoff or effluents. Consequently, it is possible to identify the source of a pollutant, or the relative contribution made by a number of sources by testing the runoff for suspected substances that might reduce water quality.

PURPOSE: To learn how to detect the pattern of surface runoff, sewage and sewage effluents, and their polluting capacities.

MATERIALS:

Map-making materials
Screw-cap water collecting bottles
Appropriate testing materials for phosphate, nitrate, coliform, detergent and others as deemed necessary

PROCEDURE: This investigation can be carried out at the school site, at a neighborhood site, or along the Indian or Banana Rivers.

1. Select a specific tract of land along, or surrounding a canal, lake,

or river. Map the area identifying all of the sources of runoff and the direction of water flow. This could be done with a carpenter's level during dry weather, but can best be accomplished immediately following a rain storm. Number, label, and classify all of the effluent and runoff sources, for example: #1 - West parking lot; #2 - Front rain gutter; #3 - Front lawn; #4 - Flamingo Road ditch; #5 - Effluent pipe; #6 - Deep well.

2. Immediately following a moderately heavy rainfall, collect water samples of each of the runoff or effluent sites. In addition, one or more samples should be taken from the river, lake, or canal itself (it would be valuable to have water samples taken here, both before and after the rain, and from a number of collecting sites).

3. Make one or more appropriate analysis of these samples for either phosphate, nitrate, coliforms, BOD, plankton, or detergent. Only make any one particular analysis if you suspect that it might be a source of one of these potential pollutants. You should, perhaps, run some samples that might not contain the suspected pollutant.

4. Make a chart showing the source of and kind of pollutant on the watershed.

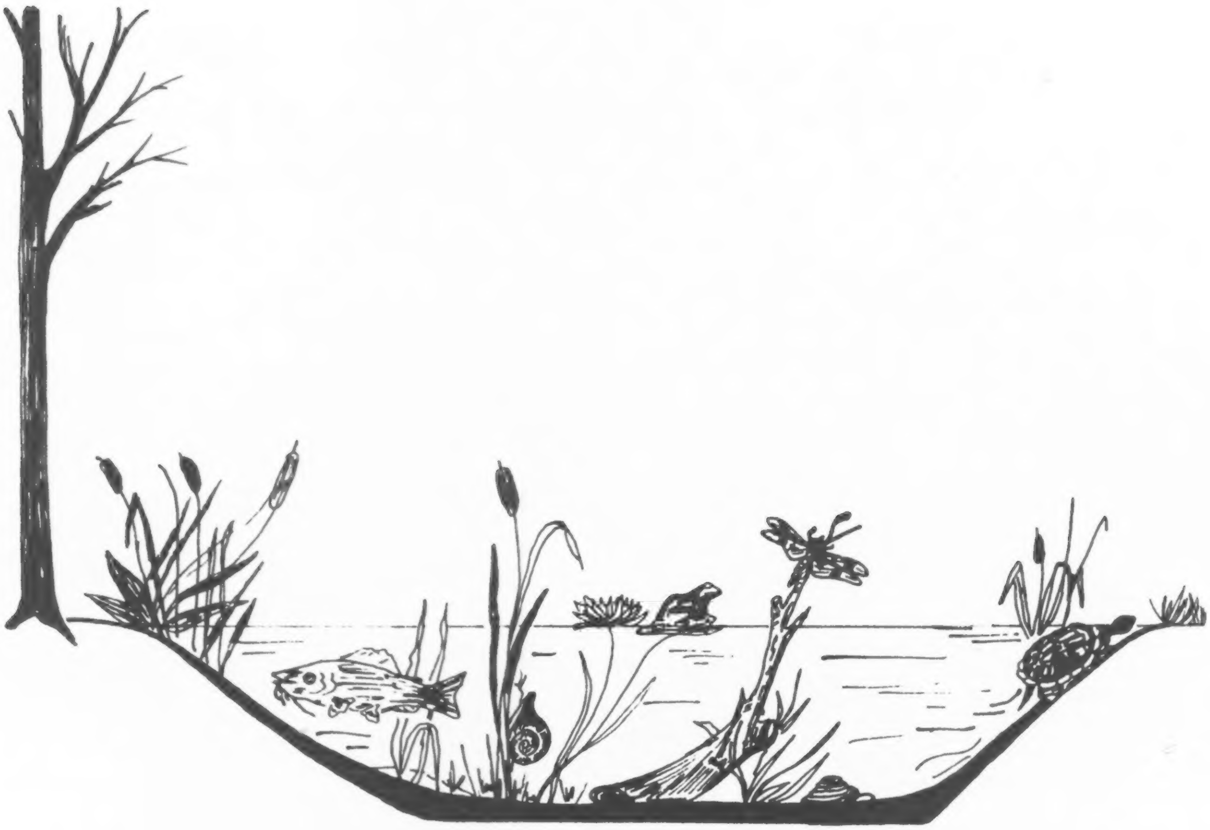
OPTIONAL: Keep samples of the water collected from each station in gallon jars. Introduce 2 small fish (Example: 2 male Gambusia 1 inch long) and a 6 inch sprig of Elodea into each jar. Keep a record of the following over a period of 2-3 weeks:

1. The exact length of the Elodea.

2. Any change in the condition of the *Elodea* or fish, e. g. parasites, color, or death.

3. Growth of algae or other plankton.

PHOSPHATE POLLUTION



Phosphates enter our natural waters, rivers and lakes from upland groves. pine woods, pastures and even our city streets. These combined with phosphates from sewage can pollute our waters through an excessive growth of algae which is not balanced by plant eating (herbivorous) aquatic species. The algae will compete with the fish for oxygen causing their death.

INVESTIGATION: AQUATIC POLLUTION INDICATORS AND INVERTEBRATE SAMPLING

BACKGROUND: A study of indicator species more accurately describes average conditions at certain points in the water body than do chemical tests.

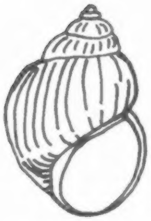
The following indicator species are responsive to dissolved oxygen levels. Therefore forms of pollution which cause oxygen depletion are indicated by the presence of characteristic organisms at certain levels of dissolved oxygen. Organic pollution such as sewage, fertilizer runoff and feed lot runoff can often be identified using the benthos method.

When studying a stream, the sampling locations would be downstream from the suspected pollution source. When studying a lake, sampling stations should radiate from the source. This technique is adapted from a procedure by A. R. Gaufin.

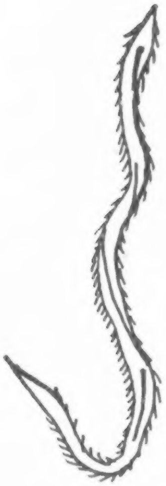
Septic Zone Indicators A septic zone is normally characterized by species adapted to live under low oxygen conditions (less than 1 ppm) or those able to secure their oxygen directly from the air.

Water bugs (Hemiptera), water beetles and even mosquito larvae are also found in septic zones but can be found in high oxygen zones as well as in equal numbers. They are poor indicators of oxygen and should not be considered.

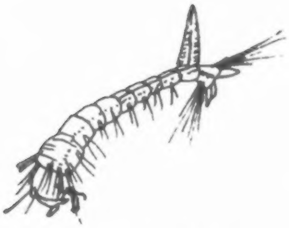
SEPTIC ZONE INDICATORS



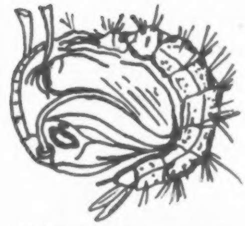
Pulmonate snail



Sludge worm



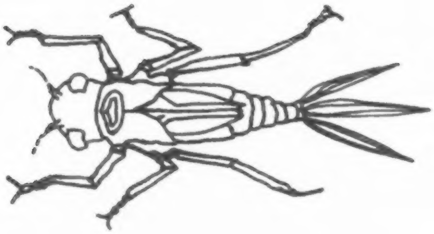
Mosquito larva



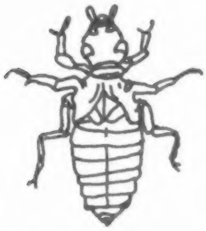
Mosquito pupa

Recovery Zone Indicators Average conditions as found in a recovery zone (downstream from septic zone) consist of lesser numbers of the more tolerant forms found in clean water, especially those having a variety of methods for securing oxygen.

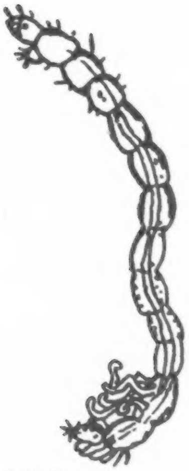
RECOVERY ZONE INDICATORS



Damselfly naiad



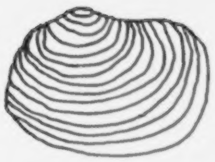
Dragonfly naiad



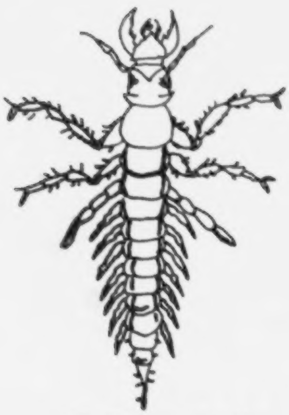
Midge larva

Clean Water Indicators Clean waters are characterized by a great variety of invertebrate communities consisting of herbivores, carnivores, and omnivores; prey and predators; lung, tracheal tube, and gill breathers. In general a population containing abundant gill breathing forms, mayflies, dragonfly nymphs and caddis flies is indicative of clean water conditions and their absence denotes the presence of pollution and/or low oxygen.

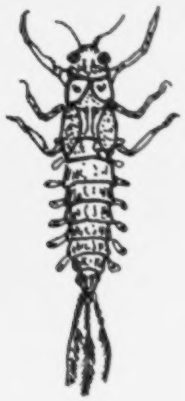
CLEAN WATER INDICATORS



Clam



Hellgrammite



Mayfly naiad

INVESTIGATION: MOSQUITO ECOLOGY AND LIFE HISTORY

BACKGROUND: The mosquito is not only a potential vector of human and animal disease, but has profound effects upon man-land relationships in our attempts to control or even erradicate one or another of the species. Florida has 67 of the world's 1500 mosquito species; of these about one dozen can be classified as severe biting pests and about six of them are real or potential carriers of human disease. These are (1) Anopheles quadrimaculatus which carries a Plasmodium (Protozoa) the causative agent of malaria; (2) Aedes aegypti which carries a virus, the causative agent of yellow fever and dengue; (3) Culex nigripalpus, a vector which carries an arbovirus which can cause St. Louis encephalitis; and other species that carry viral diseases. Malaria, yellow fever, and dengue parasites are no longer available to their mosquito vectors, consequently these diseases have banished in Florida.

In order to control mosquitoes, we have slowly limited the use of fogging and larvaciding with persistant chlorinated hydrocarbons (DDT, deldrin, and endrin). Water management is now considered the best course for permanent control; however certain aspects of these programs might prove to be inconsistent with good water conservation and wildlife management practices, that is, drainage and chemical larvaciding.

It would therefore be valuable for a student to know some of the facts concerning the ecological life-history of the mosquito. These can best be learned by rearing the mosquito through the egg, larval, pupal, and adult stages of its metamorphosis, and to acquaint the student with a variety of mosquito control measures through laboratory or field experiences. In this exercise mosquitoes will be raised in the laboratory and the larval stages will be subjected to natural, biological controls, as compared to chemical control measures.

PURPOSE: To learn to recognize the stages in the life history of the mosquito and to become familiar with chemical biological control methods for the elimination of mosquito larvae.

MATERIALS:

1. Mosquito eggs can be obtained from:

Dr. James Haeger
Entomological Research Center
P. O. Box 520
Vero Beach, Florida 32960

2. Enamel or glass dishes (13 x 9 x 2 in.) with glass plate covers.

3. Larval food: A natural infusion supplemented with a little powdered dog biscuit or see Methods for other media.

4. Eyedroppers.

5. Insecticides, larvaciding oil, *Gambusia*, dragonfly larvae or other predators.

METHODS:

1. Larval mosquitoes can be obtained from rain water puddles in nature or the larvae may be cultivated in the laboratory (see supplement #1).

2. Divide the reared or captured larvae between experimental and control porcelain pans or glass jars with a maximum amount of surface to volume ratio.

3. Add chemical larvacides, oil slicks, dragon fly larvae, Gambusia (mosquito fish) or other predator species to each of the containers. (Chemical larvacides might be obtained from the local mosquito control unit.) Set up one or more identical--save one variable--control cultures.)

4. Count the number of larvae each day. Observe the behavior of the larvae. How are they affected?

SUPPLEMENT NO. 1

Mosquito Rearing Procedures

1. Before hatching:

Remove eggs from cold box for 6-7 days and hold at 26-28° C. Barely cover the eggs with water in a small dish 1-2 in. diameter and either expose them to a slight vacum or add a few grains of baker's dry yeast. About 10-20 minutes of either treatment should result in a synchoronous hatch.

2. Larvae:

Use white enamel pans, 13 x 9 x 2 inches. Count desired number of first instar (newly hatched) larvae into each pan, add 350 ml of dechlorinated water. (It is easier to count the larvae into a dry pan by dropping many small drops with 4-6 larvae in each, from a medicine dropper.)

The pans with larvae are covered with a glass plate to cut down evaporation. The glass cover also seems to help prevent scum from forming on the surface of the water.

The larval feeding schedule is as follows:

Larval stage	Day	High diet mg/pan of 75 larvae	Medium diet mg/pan of 125 larvae	
1 instar	1*	50 Y**	100 Y	
2 instar	2	50 L		50 PL
3 instar	3	50 Y - 50 L - 50 LC	150 Y	
4 instar	4	50 Y - 50 L - 150 LC	200 Y	
4 instar	4	50 Y - 50 L - 150 LC	150 Y	

*Day of hatch

**Y - powdered brewer's yeast; L - powdered lactalbumin;

LC - ground "standard Purina laboratory chow" (passed through 40-mesh screen);

PL - powdered liver.

The high diet is about maximum that can be added without killing larvae from surface scum on the water. This gives the maximum expressivity of the developmental life history. Lactalbumin and liver powder are available from the Nutritional Biochemical Corporation in Cleveland, Ohio. The very low diet consists of one-half the high diet with 75 larvae-pan.

We use plastic scoops, made to deliver 50, 100, or 150 mg of lactalbumin, yeast, laboratory chow or liver each time they are filled. The different components of the diet are measured (with the scoops) and combined in a vial, one vial of diet-mixture for each pan of larvae. When the diet in the vial is ready to be fed, a few ml of water are added so that in 10-20 minutes the diet mixture absorbs water and becomes thoroughly wet. The food is then rinsed from the vial into the pan of larvae and sinks to the bottom rather than spreading over the surface of the water.

3. Pupae:

At 26° C and with the above feeding schedule for both 75 and 125 larvae, pupation begins on the 5th day, and on the 6th day is complete. The pupae can be collected by emptying the pans through a sieve.

The pupae are rinsed into a bowl in clean water and set in a screen cage for emergence.

4. Adults:

Mating takes place most rapidly in a cage at least 12 x 12 x 12 inches inside dimensions. The percentage of inseminated females reaches 90 to 100% in 5-6 days. Inverted vials of sucrose plugged with cotton wicks are suspended through holes in top of the cage.

As an oviposition site wrap cheesecloth (4-5 layers) around a ball of cheesecloth (2 x 2 x 4 inches). This cloth should be kept moist with either 10% sea water or Ringers solution.

5. Eggs:

A. Manipulation prior to storage:

1. Remove egg pads on Monday and Friday and date.
2. Moisten slightly and place in a vegetable crisper for 5-7 days at 26-28° C for embryo development.
3. Place crisper at 8-10° C for 2-3 days to induce dormancy of embryos.
4. Unfold cheesecloth and wash eggs into a large bowl or

rearing pan of ice water to further prevent hatching.

5. Strain eggs into a 100-mesh screen and then wash into a 3-4 inch diameter white bowl, with not more than one inch of ice water. Move the bowl in a rotary motion, causing eggs to collect at center.
6. Immediately remove eggs with a medicine dropper and place on squares of nylon cloth in 4-inch square covered plastic petri dishes prepared as follows:
    a. First place a layer of glass wool on the bottom.
    b. Over this place a rectangular piece of fiberglass screening (one-half inch longer than width of dish so that it can be tightly pressed in against the glass wool).
    c. Nine 1-inch squares of nylon cloth can be placed on the screening.
7. After the eggs are placed on the nylon cloth they should be evenly distributed with a camel's hair brush (do not leave piled up). Do not put more than 200 eggs per nylon square.

B. Storage:

1. Store a large crisper with enough depth to stand petri dishes on edge.
2. The plastic vegetable crisper should be prepared by placing a 1/2 inch layer of glass wool on bottom over which is placed a sheet of rubber (used for laboratory tops).

3. Keep the glass wool wet at all times and seal the two long sides with strips of electrician's tape.
4. Store at 8-10° C in an incubator; do not let temperature fall below 0° C.

INVESTIGATION: ANALYSIS OF NATURAL WATERS FOR PHOSPHATES

(Advanced Technique)

BACKGROUND: Phosphates (PO_4) occurring in water in the form of organic polyphosphates can be converted to orthophosphates by bacterial action. Natural waters also contain organic phosphates (as a component of the particulate suspension which can be removed by filtration) and as soluble organic molecules. The purpose of this analysis is to determine the quantity of orthophosphates in samples of water collected from sites such as drainage ditches, rivers, ponds, or the sea. Such analysis might indicate the sources of harmful amounts of this pollutant which destroys bodies of water through excessive algal growth. Too much phosphate is an indication of a "sick" river, lake, or canal.

PURPOSE: To determine the quantity of phosphate in natural water at school or community study sites. This method is more precise than the Hach method.

MATERIALS:

1. Screw cap collecting bottles
2. Millipore filter apparatus
3. Reagents:
    a) Phenolphtalein indicator
    b) Concentrated sulfuric acid solution
    c) Ammonium molybdate reagent
    d) Aminonaphtholsulfonic acid reagent
    e) Stock phosphate
    f) Standard phosphate solution (ref. Standard Methods, 1965, p. 232)
4. 2 or more Erlenmeyer flasks, 125 ml
5. A quantity of 5 ml pipettes (clean and dry)

PROCEDURE:

1. Water samples should be collected in clean screw top bottles. These samples can be stored until they are needed, by the addition of 5 ml of chloroform/liter sample.

2. Natural water samples should be carefully filtered to remove particulate materials which will otherwise interfere with the colorimetric analysis. Vacum filtration with appropriate millipore filters would be most effective.

3. Color development:
   a) Pipette 50 ml of the solution to be tested (unknown) into a clean dry 125 ml Erlenmeyer flask.
   b) Add 2.0 ml of reagent c to the test solution above and mix.
   c) Add 2.0 ml of reagent d to the same solution above and mix.
   d) After 5 minutes read the sample at 690 mm (or 650 mm if necessary) on the colorimeter.

4. Before reading the unknown sample a "reagent blank" must be prepared as follows:
   a) Pipette 50 ml of distilled water into a 125 ml flask.
   b) Add 2.0 ml of reagent b.
   c) Add 2.0 ml of reagent d to the solution above and mix.
   d) Using the "reagent blank" adjust the colorimeter to 100% transmittance or zero optical density.

5. Phosphate Standards:
   a) Make a series of dilutions of the phosphate standard solution;

full strength, 3/4 strength, 1/2 strength and 1/4 strength. (All final volumes should be the same, 60 ml.)

b) Develop the color as indicated in section 3 above.

c) Plot the data, optical density vs. mg of phosphate on regular graph paper or mg of phosphate vs. transmittance on semi-logarithmic graph paper.

QUESTIONS:

1. Why is the filtration of water samples necessary for accurate phosphate (PO_4) analysis?
2. How is the color of the reaction mixture related to the color of the light (690 mm) used in this analysis?
3. How could washing laboratory glassware with detergent cause an inaccurate PO_4 analysis?
4. How does a high phosphate content in water cause a lowering of the oxygen content?
5. List the sources of the phosphate found in natural waters.

REFERENCES:

Standard Methods for the Examination of Water & Sewage, 12th ed., American Public Health Association, Inc. (1965) 1740 Broadway, New York, N. Y. 10019.

Colormetric Precedures and Chemicals for Water and Wastewater Analysis With Calibration for the Bausch & Lomb Spectronic 20 Colorimeter. Hach Chemical Co., Box 907, Ames, Iowa 50010.

APPENDIX A

SUBJECT MATTER CROSS REFERENCE

LIFE NATURE LIBRARY

Ecology By Peter Farb
Time, Inc.

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1. Introduction	Introduction 7, 9 16
2. The Nutrition Web (Producers & consumers)	How a food chain may begin 106-109; Parasites as predators 110-111; Food Pyramid 37; Coexistence in community 35-54
3. Community (Interaction of species symbiosis, niches, structure)	Coexistence in the community 35-53; Mutual aid-or Tooth and claw 95-119; Commensalism 113; Mutualism 114
4. Ecosystem (Interaction of biota & physical environment)	Heron in separate niches 40-41; Niche 16; Community and habitat 15
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THE SEASHORE
William H. Amos
Golden Press

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9. Man vs. Nature Pollution Environmental Destruction	Man comes to the shore 188, 191; Mistakes by man 191-197; Vanishing seashore 212-213

10.	Behavior (Territoriality, reasoning, imprinting)	Diurnal animals 55; Biological clocks 52; Nocturnal " 55 31, 35-39; Migration 177, 184, 171-175, 172-173
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ODUM'S ECOLOGY

Holt, Rhinehart, Winston

	TOPICS	
1.	Introduction	
	The Nutrition Web (Producers & consumers)	Chap. 3, 37-52; Chap. 5, 66-67; Chap. 4, 53-69
2.	Community (Interaction of species symbiosis, niches, structure)	Chap. 2, 25-28; Chap. 6, 107-109
3.	Ecosystem (Interaction of biota & physical environment)	Chap. 2, 7-36;
4.	Population	Chap. 2, 17-20; Chap. 6, 91-95; 88-109
5.	Biomes	Chap. 7, 112-135
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7.	Hydro-Ecology	
	Marine	Chap. 7, 112-118
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8.	Man vs. Nature Pollution Environmental Destruction	Chap. 5, 75
9.	Behavior (Territoriality reasoning, imprinting)	Chap. 6, 93-100
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LIFE OF THE MARSH
William A. Niering
McGraw-Hill

TOPICS

1.	Introduction	
2.	The Nutrition Web (Producers & consumers)	The flow of energy 61-71, 82, 96-101, 103, 156, 168, 169, 173, 186, 223, 224
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LIFE OF THE POND

William H. Amos
McGraw-Hill

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APPENDIX B

FIELD TRIP INFORMATION AND TAXONOMIC KEYS

WUESTHOFF NATURE PARK

LOCATION: Barna and Route 50 in the southwest section of Titusville, Fla.

BACKGROUND: This north Brevard park has a great variety of living communities located in a relatively small (20 acre) area. The habitats include a pond, canals, grassy fields, hammock, a bayhead, and some sand pine scrub-six in all.

The park has a large parking area, picnic benches, restroom facilities, and a handicraft cottage. Tours are arranged through the Parks and Recreation Office in Titusville. (269-8354)

PLANT & ANIMAL COMMUNITIES:

A. Hammock

This dense hardwood forest is dominated by the following trees & shrub species: laurel oak (*Quercus laurifolia*), stiffcornel dogwood (*Cornus stricta*), live oak (*Quercus virginiana*), laurel cherry (*Prunus carotiniana*) and cabbage palm (*Sabal palmetto*). The presence of many epiphytic ("air plants") species is characteristic of the hammock. This portion of the forest grades almost imperceptably into the bayhead.

B. Bayhead:

A bayhead is dense swamp typically not along a stream or river. The roots of the trees are often exposed and are interwoven into a tangled mass. The floor of the forest is often inundated during the rainy season, but local canals have initiated some drainage at Wuesthoff. The largest and dominant tree is the sweet bay (*Magnolia virginia*). Elderberry (*Sambucus Impsoni*) is common on the unshaded edges of the bayhead. Elderberry fruit furnishes food for many birds. Numerous red maple (*Acer rubrum*) and loblolly-bay (*Gordonia lasianthus*) trees are also present. The loblolly-bay is always conspicuous in summer due to the presence of beautiful large white flowers.

C. Sand Pine Scrub:

As the nature trail leaves the bayhead you will find that the elevation increases a few feet into a clearing. This is a sand pine scrub community that has been greatly modified by man; however, a few of the typical species can be seen. Only three species of trees seem to grow successfully in this habitat; these are the sand pine (*Pinus clausa*), the scrub hickory (*Carya floridana* , and sand live oak (*Quercus geminato*). Woody shrubs unique to this old dune association are myrtle oak (*Quercus mertifolia*), and rosemary (*Ceratiola ericoides*).

D. Pond & Canal:

Only a few emergent aquatic plants were identified here. Along the edge of the pond buttonbush (*Cephalanthus occidentalis*) is common. This woody shrub is easily identified by the presence of a globose head (composite) of flowers. Along the water's edge is pickerel weed (*Pontedera cordata*) with spikes of blue flowers. Floating on the water's surface is duckweed, a minute flowering plant.

SIMPLE KEY TO A FEW SPECIES OF WUESTHOFF NATURE PARK TREES

A. A tree -- B
A' A woody shrub ---------------------------------- not in key
A" A vine -- not in key

B. A palm tree-- Cabbage palm
B' Not a palm, pinnate venation -------------------- C

C. Leaf simple with 3-5 lobes, red petiole ----------- Red maple
C' Leaf compound, 3-5 serrate leaflets, large nuts----- Scrub hickory
C" Leaf not lobed or compound ---------------------- D

D. A pine, needle - like leaves ---------------------- E
D' broad leafed tree ------------------------------- F

E. Needles 3-4 inches long ------------------------ Sand pine
E' Needles over 5 inches long --------------------- Slash pine

F. Rough bark with hard but brittle entire leaves ------ G
(Bark with few or no lichens)
F' Relatively smooth bark -------------------------- H

G. Large hammock trees with obovate entire leaves ---- Live oak
G' Small scrub trees with elliptic-lanceolate entire leaves ---- Sand live oak
G" Leaves ovate with a serrate margin --------------- Fla. elm

H. Leaves obovate, usually a shrub-like oak, sandy soil-- Myrtle oak
H' Leaves not obovate moderate to large trees --------- I

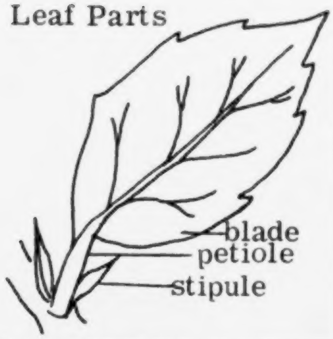
I. Leaf margins somewhat serrate ------------------ J
I" Leaf margins entire, leaves alternate ------------- K

J. Flowers white, 3 in. in diameter, leaves green on both sides, canopy tree -------- Loblolly bay
J' Flowers very small, understory tree ------------- Laurel cherry

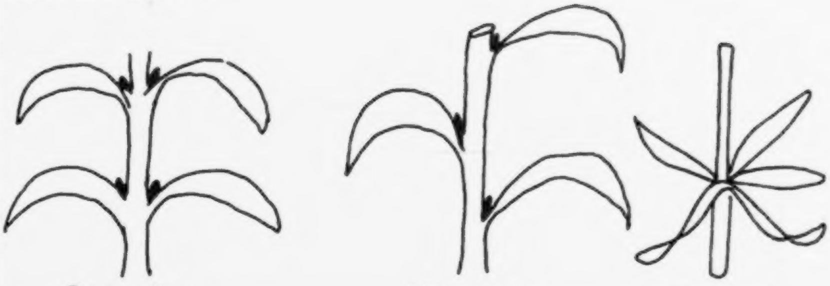
K. Leaf very silvery beneath, elliptical --------------- Sweet Bay
K' Leaf not silvery white beneath -------------------- L

L. Dark purplish brown bark, aromatic leaves --------- Redbay
L' Bark, smooth light in color with large lichen patches, elliptical leaves plyable; acorns in season ---------- Laural oak
L" New potato-like bark, understory tree ------------ Stopper

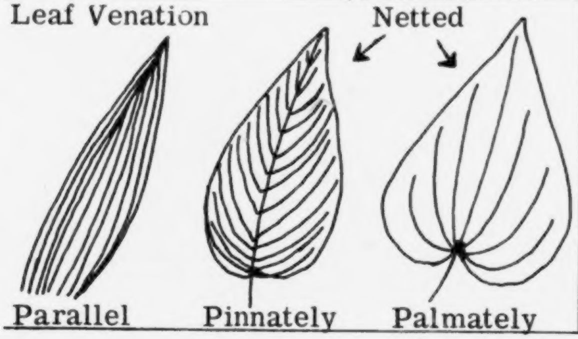
Leaf Parts


blade
petiole
stipule

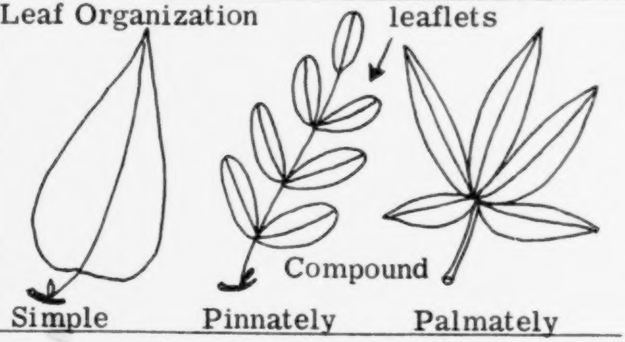
Leaf Position


Opposite
Alternate
Whorled

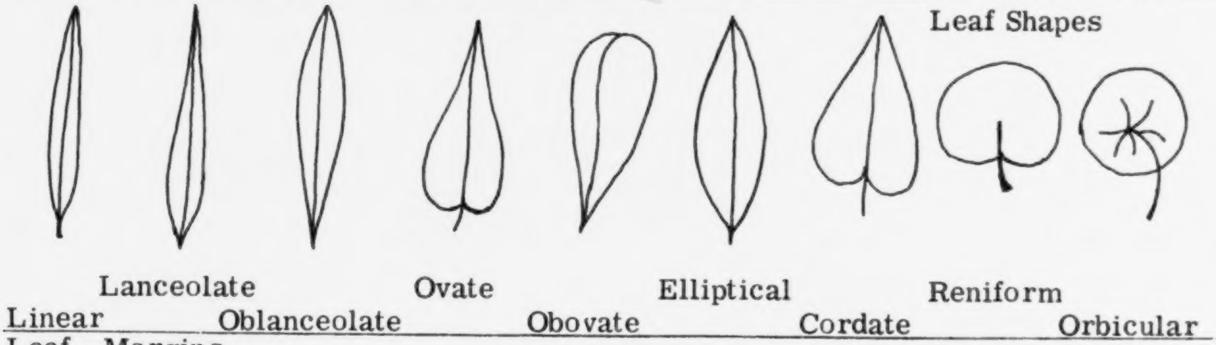
Leaf Venation


Netted
Parallel
Pinnately
Palmately

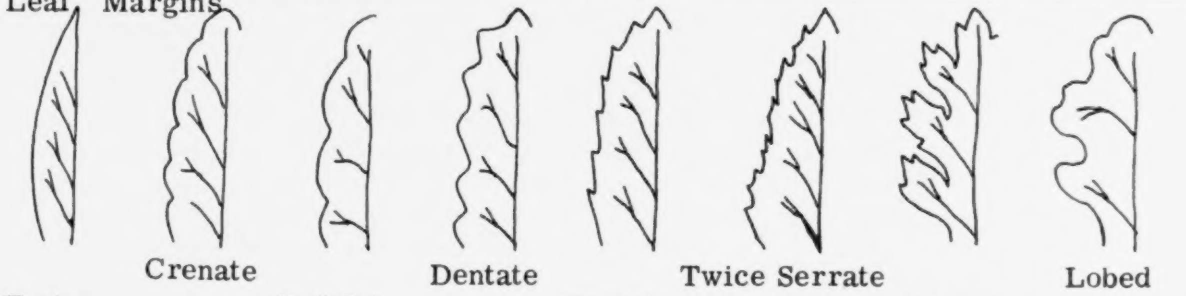
Leaf Organization


leaflets
Compound
Simple
Pinnately
Palmately

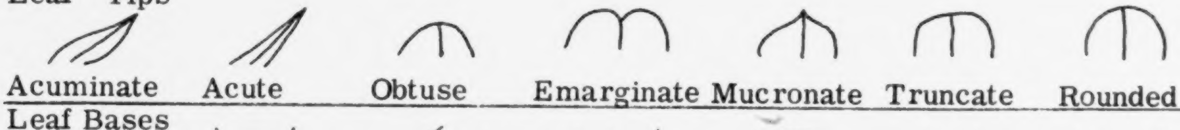
Leaf Shapes


Lanceolate
Ovate
Elliptical
Reniform
Linear
Oblanceolate
Obovate
Cordate
Orbicular

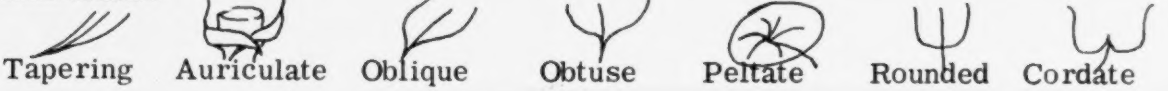
Leaf Margins


Crenate
Dentate
Twice Serrate
Lobed
Entire
Undulate
Serrate
Incised

Leaf Tips


Acuminate
Acute
Obtuse
Emarginate
Mucronate
Truncate
Rounded

Leaf Bases


Tapering
Auriculate
Oblique
Obtuse
Peltate
Rounded
Cordate

FIELD USE OF THE CAMERA

Film records of observations are one of the best teaching aids possible in relating field experiences to class work. Without too much trouble nor expense the teacher can provide this learning tool for the class.

There are several types of cameras used for this type of activity. They fall into several categories.

1. Non-Range Finder - These models have no optical range finder and the subject to camera distance must be estimated or measured and then set on the camera.

2. Range-Finder Model - A small viewer window is calibrated to allow the photographer to set the subject distance by focusing the camera. This may be a split image that merges into one when properly focused.

3. Single Lens Reflex (SLR) - The photographer views the subject through the lens by way of a swing away mirror. This allows excellent focusing and composition. Just before the film is exposed the mirror swings away. Probably the most versatile field camera as it usually accepts a number of accessories: Film size is most often 35 mm. as larger sizes are very expensive.

4. Twin Lens Reflex (TLR) - This type has two lenses - one for viewing and one for transmitting the image to the film. The film size is usually 120, 127, or 220. Focusing is extremely accurate and composition is easily accomplished.

5. View Cameras and Press Cameras - The subject is viewed through the same lens as the picture is taken. Film size varies from roll sizes to 8 x 10 inch cut film. These are excellent cameras but are heavy and bulky.

Film chosen will depend on the camera and type of photography to be done. Generally speaking one type of film should be chosen and used exclusively until the photographer is well acquainted with his camera.

Many film speeds are much higher than a few years ago thus reducing the exposure time needed. Some new films are very slow and produce extremely well defined non-grained enlargements.

Generally, most will use a black and white film such as Kodak's Plus X with an A.S.A. of 125 and a color film like Ektachrome with an A.S.A. of 160 for color slides. By using two basic types of film - one black and white and one color - and not changing to other films the photographer will become familiar with his basic film and what it is capable of doing under all conditions.

Most pictures taken in the field tend to encompass too much area. If the end product is to be a slide, get close enough to the subject so that it will fill the frame of the picture. Some subjects are so small that the camera is not capable of focusing close enough to get a good reproduction. In this case one of three methods are used:

(1) Close up lens - Attachment lens that slip over the camera lens and shorten the focal length.

(2) Extension tubes - Tubes that fit between the lens and camera to allow close-up focusing. Only for cameras with removable lenses.

(3) Bellows attachment - An adjustable extension device that fits between the lenses and the camera.

(4) Marco or close-up lens - Replaces normal lens and allows close focusing.

In close-up photography motion is a problem. Tripods eliminate most camera motion and a cable release will help even more. Subject motion is a problem to be dealt with in each individual case. Wind screens, sun shades, glass partitions, and other aids are a necessary part of the photographers equipment.

Pictures used in the classroom can be reused year to year and serve as a basis for comparison as the subject changes by growth, erosion, fire, etc.

MEASURING THE HEIGHT OF A TREE

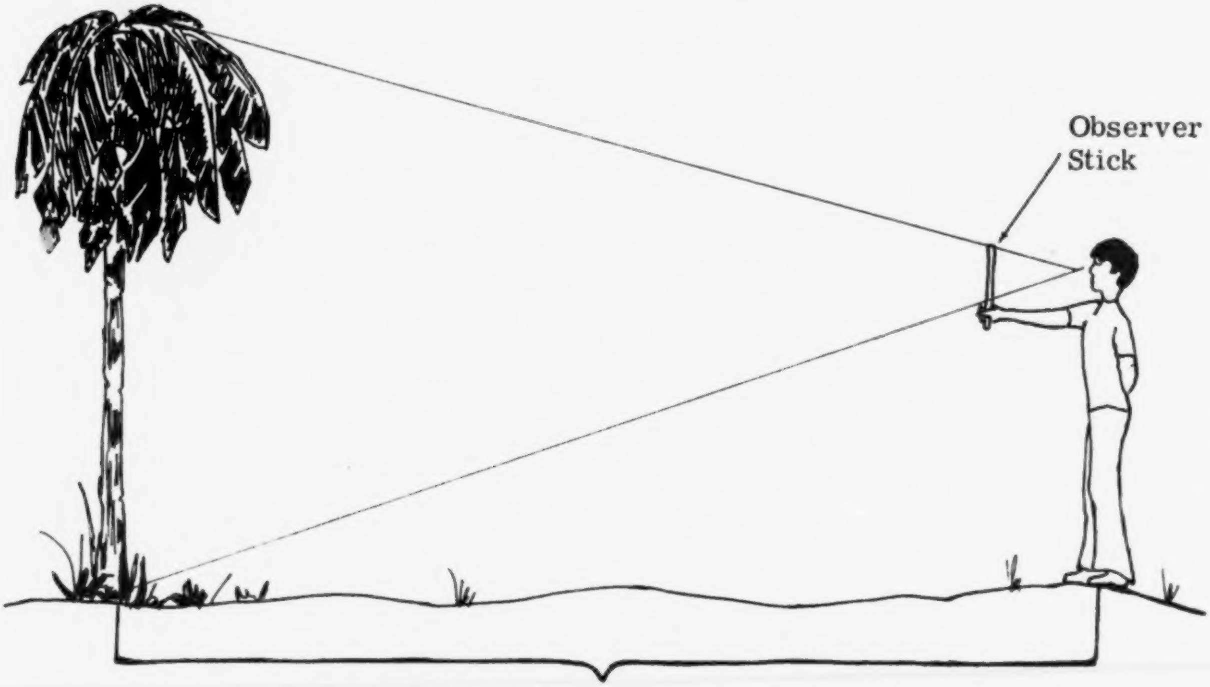
Observer should:

1. Select a stick of the same length as the distance from his eye to his knuckles when his arm is held parallel to the ground.

2. The stick is held vertically in such a manner as to form a right angle with arm.

3. Observer walks backward from the tree until he is sighting across the top of the stick at the top of the tree and the base of the tree across his hand.

4. The distance from the tree to where observer is standing is equal to the approximate height of the tree.



This distance is the approximate height of the tree

FIRST AID

There are many small incidents on field trips that will require First Aid attention. These should present no major problems if good common sense is used. The material that follows will be of use as a guide line. A good First Aid course should be a part of every field teacher's background. The Red Cross will provide courses at your request.

Prevention is always the best part of any First Aid - keeping this in mind students should be:

1. Warned about sharp objects in shirt pockets and on belts.
2. Advised about proper footwear.
3. Informed in advance about a trip.
4. Shown how to recognize poisonous plants.
5. Told about the habits and dangers of animals.

Some possible sources of injury and treatment thereof are as follows:

(1) INSECT STINGS

Since most stings are acid, application of a base to the sting area will help. Make sure that the stinger is no longer in the wound. Apply a paste made of baking soda and water to affected area. Watch for signs of allergic reaction to the sting. If severe swelling, pain or respiratory distress occurs consult a physician as soon as possible.

(2) PLANT POISONING

The most common is poison ivy which contains the oleoresin urushiol. This hazard is greatest during spring and summer months.

After contact, the skin becomes red and small blisters appear, followed by itching. As blisters break, the area may increase in size.

Soap and water followed by rubbing alcohol applied to the wound will minimize the reaction. Calamine lotions may relieve the itching.

(3) SPRAINS AND FRACTURES

A. Sprains

Sprains can be prevented by wearing proper footwear, making sure footwear is laced properly. No horseplay when in the field and at all times WATCH YOUR STEP!

If, however, sprains should occur in field work they should not be ignored as they can become progressively worse. They occur when a moveable joint is pushed past its normal stopping point and damages the soft surrounding tissues. If available, immediate applications of cold compresses will reduce pain and swelling. After swelling stops apply warm compresses.

A sprained member should not be used unless it is necessary. Bandaging a sprained ankle and then walking does not help the sprain, in fact, it may prolong the recovery time.

B. Fractures

Fractures, or broken bones, are classed as open or closed. An open fracture is associated with a skin wound extending from the surface to the fracture. A closed fracture is simply a broken bone.

Prevention follows the same lines as above in sprains. Falls cause the majority of all fractures.

In order to protect an injured person a fracture must be immobilized by the best means available as soon as possible. Magazines, boards, sticks, blankets may be used to provide support for a fracture. Swelling accompanies a fracture. Therefore the binding on the support material must be checked periodically to insure that circulation is not impaired. Medical attention is necessary.

(4) WOUNDS

Minor wounds should be cared for immediately to prevent infection. Soap and water are the best prevention available to the first aider. The wound should be washed and bandaged.

Severe wounds obviously will need medical attention as soon as possible. Bleeding should be stopped immediately by:

1. Direct pressure on the wound along with elevation. OR
2. Vessel pressure near the wound as required (brachial artery (arm) or femoral artery (leg). OR
3. Tourniquet - used as a last resort - once applied, do not loosen. Let a physician remove the tourniquet.

(5) ARTIFICIAL RESPIRATION

Stoppage of breathing requires immediate action to maintain life. The best method, always available, is mouth to mouth resuscitation. Check the victims mouth for obstruction. Tilt the head back so that the jaw "juts" out, gently pinch shut the victim's nose, cover his mouth with yours, and blow gently until the chest shows expansion. If no expansion occurs recheck the mouth for obstruction, re-position the head and try again. As soon as the chest starts to rise or expands, remove your mouth and let the air escape from the victim's lungs. Then cover his mouth again and blow.

Keep this procedure going at the rate of 12 times a minute until the victim recovers or medical help is obtained. If the victim recovers - watch him carefully until medical attention is available as he may suffer a relapse.

(6) SNAKE BITE

When struck by one of the pit vipers (rattler, cottonmouth or copperhead) a pocket of venom is injected through each of the fangs while in the flesh. This toxin affects the circulatory system but very few victims die, contrary to popular belief. Secondary infections are very common in snake caused wounds.

Pain occurs immediately along with swelling and discoloration. The victim may experience shortness of breath, weakness, nausea and a rapid pulse rate.

The victim must relax and be as calm as possible. Affix a constriction band above the point of injury. This band is to slow surface circulation and is not a tourniquet. There should be some blood ooze from the wound. Make sure this band stays above the swelling, which will spread as time goes on after the injury.

If the circumstances warrant it, use a sterile blade to make 1/4" long incisions over the fang mark in the form of a cross or an X. Cuts across the limb should be shallow, those that are longitudinal can be longer.

Apply suction to the area to remove as much venom as possible. Continue for an hour or until medical assistance is reached. Cold applications are helpful for pain and may slow absorption.

RECOMMENDATION:

A snake bite kit is provided to each class who will be engaged in Field Activities. It is important that the teachers and any other adult leaders who may participate in Field Trips study the instructions closely and perhaps simulate and practice the necessary steps which would be involved in the treatment of snake bites. Bear in mind that involvement with a snake in the field usually results in varying degrees of hysteria among the students, and it is vitally necessary for the teachers and other adults to remain calm and be able to systematically proceed with the recommended treatment as outlined in the Snake Bite Kit Instructions.

A Field Key to the Freshwater Algae

1 - Living in or upon animals 2
2 - Not living in close association with animals 5

    2 - On shells of snapping turtles, filamentous, tufted . . . . *Basicladia*
    2 - Not on turtle shells 3

3 - Forming a hard green coating on snail shells . . . . . . *Gongrosira*
3 - Not on snail shells 4

    4 - In old egg masses of the salamander Ambystoma, dark green . . . . *Oophilia*
    4 - Living within green Hydra or green Planaria . *Zoochlorella*

5 - Aquatic, submerged or nearly so 14
5 - Not aquatic, on soil, rocks, wood or bark 6

    6 - On soil or rocks 8
    6 - On wood or bark 7

7 - On bark or shaded side of tree trunks or on weatherbeaten siding of old buildings . . . . *Protococus*
7 - On rotting logs or pilings, pale green . . . . *Hormidium*
Stichococus

    8 - On rocks 9
    8 - On soil 10

9 - Algal mass orange or reddish, on "Dry" cliff or quarry face . . . . *Trentepohlia*
9 - Algal mass dark olive to black, slimey *Oscillatoria*

    10 - Algal mass filamentous, often in greenhouses 11
    10 - Algal mass not filamentous or felt-like, varied locale 13

11 - Dark green, felt-like, coarsely branched . . . . *Vaucheria*
11 - Yellow green, tawny or olive brown 12

    12 - Yellow green, filmy . . . . *Hormidium*, *Stichococcus*
    12 - Tawny or olive brown, velvet or felt-like . . . . *Scytonema*

13 - Algal mass jelly-like, spherical or in expanded sheets . . . . *Nostoc*
13 - Algal mass globular, 1-2 mm. in diameter, shining with white flakes . . . . *Botrydium*

14 - Running water, shore lines or in spray, attached 15
14 - In standing or very slow moving water, attached or not 28

15 - Water cold (as in early spring or spring fed) 16
15 - Water warm (late spring or summer months) 24

16 - Algal mass filamentous 17
16 - Algal mass not filamentous 23

17 - Algal mass a felt-like mat . . . . *Vaucheria*
17 - Not a felt-like mat 23

18 - Plants not branched 19
18 - Plants much branched (bushy) 20

19 - Plants short, slippery, bright green . . . . *Ulothrix*
19 - Plants nodulose, cartilagenous, olive . . . . *Lemanea*

20 - Plants embedded in a jelly-like mass 21
20 - Plants not gelantinous, coarse, often in very swift water . . . . *Cladophora*

21 - Olive-green to red-purple . . . . *Batrachospermum*
21 - Brilliant green 22

22 - Gelatinous mass soft, indefinite, long lateral branches . . . . *Stigeoclonium*
22 - Gelatinous mass firm, definite, short lateral branches at right angles to main axis . . . . *Draparnaldia*

23 - Colony saccate, membraneous or tubular, pale green and gelatinous . . . . *Tetraspora*
23 - Colony gelatinous, brown, amorphous, spreading over rocks . . . . *Diatoms*

24 - Filamentous, branched or not 25
24 - Not filamentous, green incrustraction rocks and sticks . . . . *Chlorotylium*

25 - Filaments branched 26
25 - Filaments not branched or only at base 27

26 - Algal mass a felt-like mat . . . . Vaucheria
26 - Algal mass not felt-like, coarse,
bushy, in rapid water . . . . Cladophora

27 - Filaments short (to 1 inch) nodulose, olive,
in turbulent water . . . . Lemanea
27 - Filaments long (to 2 feet) silky, green, tough . . . . . Rhizoclonium

28 - Cold water, early spring 29
28 - Warm water, late spring, summer, early fall 34

29 - Woodland pools, leaf-litter bottom 30
29 - Open ponds, lake margins, pools, cut-offs 31

30 - Algal mass yellow-green, filamentous,
silky . . . . Tribenoma
30 - Green motile spheres visible in glass
jar of water . . . . Volvox

31 - Plants tree-like, calcareous, attached
to bottom . . . . Chara, Nitella
31 - Filamentous 32

32 - Attached to dead sticks, weeds, grasses . . . Oedogonium
32 - Free floating 33

33 - Brilliant green, slippery, ends of mass curling
when held aloft . . . . Spirogyra
33 - Bright to light green, less slippery, ends of mass
not distinctly curling . . . . Zygnema, Mougeotia

34 - Temporary bodies of water 35
34 - Permanent or semi-permant bodies of water 38

35 - Bird baths, urns, shoreline pools, reddish scum
on sides and bottom . . . . Haematococcus
35 - Puddles, cow tracks, ruts, pigpen ponds 36

36 - Bluegreen, olive, black, often on mud,
slimey, membranaceous . . . . Oscillatoria
36 - Green or red, living in, or on the water 37

37 - Forming red or green scum on surface of water . . . . *Euglena*
37 - Water colored uniformly green throughout . . . . . . *Chlamydomonas*

38 - Aquaria, bottles and culture dishes in
the laboratory or greenhouse 39
38 - Ponds, lakes, lake margins, "cut-offs" 40

39 - As a green film against glass walls . . . . *Chlorella*
39 - Water uniformly green throughout *Scendesmus*, *Ankistrodesmus*

40 - On wet soil at margins 41
40 - Aquatic, floating or submerged 42

41 - Globular, gelatinous colonies . . . . *Nostoc*
41 - Blue green, olive, black, slimy
membranaceous colonies . . . . *Oscillatoria*

42 - Submerged and attached 43
42 - Free floating 46

43 - Plants tree-like, calcareous, growing on bottom
mud, submerged from 1 - 30 feet . . . . *Chara*, *Nitella*
43 - Not tree-like, not growing from bottom mud 44

44 - Filamentous, attached to weed and
grass stems . . . . *Oedogonium*
44 - Not filamentous 45

45 - Small hemispherical or branched gelatinous
colonies, attached to weeds and sticks . . . . *Chaetophora*
45 - Flat green discs often attached to dead
cattail or water lily leaves . . . . *Coleochaete*

46 - Algal mass forming a net . . . . *Hydrodictyon*
46 - Algal mass not net-like 47

47 - Algal mass a tough membranaceous, paper-like
sheet, green to olive-green . . . . *Lyngbya*
47 - Algal mass filamentous, not membranaceous, green 48

48 - Filaments coarse, branched, not slippery 49
48 - Filaments silky, unbranched, slippery 50

49 - With scattered, dark swollen areas . . . . *Pithophora*
49 - Without dark, swollen areas . . . . *Cladophora*

50 - Bright green to yellowish, if green, very slippery, ends of mass curling if held aloft . . . Spirogyra

50 - Green to yellowish, only slightly slippery, ends of mass not curling . . . . Zygnema, Mougeotia

A Key to the Initial Separation of the More Common Plankton Organisms

1 - No chlorophyll present, unless through ingestion 8
2 - At least some chlorophyll present 2

2 - Pigments not in plastids . . . . Cyanophyta
2 - Pigments in one or more plastids 3

3 - Cellwall of over-lapping halves and distinctly sculptured . . . . Bacillariophyta
3 - Cellwall not of over-lapping halves, or if so, then not sculptured 4

4 - Pyrenoids present; color usually bright green . . . . Chlorophyta
4 - Pyrenoids absent; color green, yellow-green or yellow brown 5

5 - Bright green, motile, usually with one anterior flagellum . . . . Euglenophyta
5 - Yellowish to brownish, motile or not 6

6 - With a distinct lateral groove, motile . . . . . . Dinophyta
6 - Without a lateral groove 7

7 - Seldom motile; unicellular, colonial or filamentous . . . Xanthophyta
7 - Motile, unicellular or colonial . . . . Chrysophyta

8 - Unicellular, naked or enclosed in a smooth or sculptured shell 9
8 - Multicellular; body usually with a distinct exoskeleton 11

9 - Amoeboid; sometimes with shell, no cilia or flagella . . . . Ameboid Protozoa
9 - Actively motile; never with shell; cilia or flagella obvious 10

10 - Body more or less covered by short cilia; movement "Darting" . . . . Ciliate Protozoa
10 - Body with one or more flexible, whip-like flagella; movement "continuous" . . . . Flagellate Protozoa

11 - Shell bivalved (clam-like) 12
11 - Shell not composed of two halves 13

12 - With distinct head anterior valves . . . . Cladocera
12 - No head anterior to valves . . . . Ostracoda

13 - Usually microscopic; body extended into a tail or foot with one or more toes . . . . Rotifera
13 - Usually microscopic (if mature) 14

14 - Appendages bilateral; head not prominent . . . . Copepoda
14 - Appendages unilateral; head prominent . . . . Phyllopoda

APPENDIX C

FILMS BY TOPICS

THORNE FILM LOOPS

Purchased and earmarked for ecology

572	Rocky Coast Environment
572-1	High Tide Zone
572-2	High Tide Zone
572-3	Low Tide Zone
573	Sandy Beach Environment
577	Obelia
587	Fairy, Clam, and Tadpole Shrimp
588	Dragonfly
591	Mayflies
592	Pipevine Swallowtail Life Cycle
599	Hydra
501	Hydra
505	Tapeworm
508	Land Snail
518	Starfish
549	Actinosphaerium
550	Spirostomum
551	Didinium
552	Stentor
590	Mosquito Life Cycle
593	Horse clam
598	Ladybird Beetle Life Cycle
542	Euglena, Part I

Purchased through C & D Audio-Visual Supply
Box 5116 - 1690 S. Rio Grande
Orlando, Florida 32805

THE NUTRITION WEB
(Producers & Consumers)

Although no film recommendations are available at this time, this page is included to provide a location to list future references as they are revealed or new reference sheets are issued to up-date this initial pilot material.

Community
(Interaction of Species Symbiosis, niches, structure)

	Title	Color/B. W.	Time	Level
8-298	Nature's Plan.	C	15	J. S.
8-662	Everglades... Balanced Community	C	11	J. S.
12-327	Prowler of the Everglades	C	32	E. J.
4-574	Grass-Blade Jungle	C	11	E. J.
12-223	Beaver Valley	C	32	E. S.
8-98	Life in the Sea	C	11	E. S.
12-193	The Desert	B	22	E. S.
8-294	Temperate Deciduous Forest	C	17	E. S.
4-235	Water Birds	B	11	E. S.
4-748	Life in an Oasis - North Africa	C	11	E. J.
4-744	Life in Hot Dry Lands - California	C	11	J. S.
8-182	Life in the Desert - North America	C	11	E. S.
8-310	What is Ecology	C	11	S.
3-296	The Community	C	11	S.
4-327	Life on a Dead Tree	C	11	E. J.
8-307	The Cave Community	C	13	J. S.
4-676	Wild Flowers of the Field/Meadow	C	11	E. S.
8-657	Coniferous Forest Biome	C	15	P. S.
8-53	Life in the Grasslands	C	11	E. S.
8-98	Life in the Sea	C	11	E. S.
4-514	Life Along the Waterways	C	11	E. J.

Ecosystem
(Interaction of Biota &
Physical environment)

	Title	Color/B. W.	Time	Level
8-688	Balance of Life and The Space Age	C	14	E. S.
8-581	Distribution of Plants and Animals	B	16	E. S.
8-310	What is Ecology	C	11	S.
4-748	Life in an Oasis - North Africa	C	11	E. J.
4-572	Grass-Blade Jungle	C	11	E. J.
8-585	Grassland	B	17	E. S.
8-662	Everglades... Balanced Community	C	11	J. S.
B. F. A.	American Woodlands	C	9 1/2	J. S.
4-498	Altered Environment			

Population

	Title	Color/B. W.	Time	Level
8-586	Population Ecology	C	19	J. S.
4-51	Life Along the Waterways	C	11	E. J.
12-223	Beaver Valley	C	32	E. S.
12-327	Prowler of the Everglades	C	32	E. J.
8-298	Nature's Plan	C	15	J. S.
McGraw-Hill	Standing Room Only	C	25	J. S.
	Cities of the Future	C	25	J. S.
	Autos Everywhere	C	25	J. S.

Biomes Zoogeography	Title	Color/B. W.	Time	Level
8-53	Life in the Grasslands	C	11	E. S.
8-657	Coniferous Forest Biome	C	15	P. S.
8-581	Distribution of Plants/Animals	B	16	E. S.
12-193	The Desert	B	22	E. S.
8-294	Temperate Deciduous Forest	C	17	E.S.
4-744	Life in Hot Dry Lands - California	C	11	J. S.
8-182	Life in the Desert -North America	C	11	E. S.
8-585	Grassland	B	17	E. S.
12-327	Prowler of the Everglades	C	32	E. J.
12-284	Nature's Half Acre	C	33	E. S.
8-270	Yours is the Land	C	21	E. S.
McGraw-Hill	Life in Parched Lands	C	30	J. S.
The Estuary	Bureau of Comm. Fishing Arlington, Va.	C	30	J. S.
Imperial Film	Co. Pond, Seashore, Forest, Desert	Strips w/records		
Thorne				
572	Rocky Coast Environment			
572-1	High Tide Zone 1			
572-2	" " " 2			
572-3	Low " "			
573	Sandy Beach Environment			

Succession

8-297 Succession -
Sand Dune to Forest

Aquatic Ecosystems
Fresh Water

	Title	Color/B. W.	Time	Level
8-575	Conserving Our Natural Resources	C	11	E. S.
8-662	Everglades. . . Balanced Community	C	11	J. S.
8-418	Water's Edge	C	12	E. S.
8-301	Simple Plants - The Algae	C	18	J. S.
8-191	Fresh Water Pond	C	14	E. J.
4-467	Environment Survival - Trout	C	10	E. J.
4-51	Life Along the Waterways	C	11	E. J.
12A-225	One Day at Teton Marsh	C	23	E. J.
12B-226	One Day at Teton Marsh	C	24	E. J.
McGraw-Hill	Water - Old Problems - New Approaches	C	30	J. S
Thorne				
590	Mosquito Life Cycle			
542	Euglena			
552	Stentor			
551	Didinium			
550	Spirostomum			
549	Actinosphaerium			
501	Hydra			
592	Caddis Flies			
591	May Flies			
588	Dragon Fly			
587	Fairy, Clam and Tadpole Shrimp			
577	Obelia			

Aquatic Ecosystems
Marine

	Title	Color/B. W.	Time	Level
12A-314	Search in the Deep I	C	27	P. S.
12B-315	Search in the Deep II	C	27	P. S.
8-98	Life in the Sea	C	11	E.S.
8-539	Life of the Oyster	C	11	J. S.
8-570	Secrets of the Underwater World	C	16	J. S.
12A-312	Coral Jungle I	C	27	P. S.
12B-313	Coral Jungle II	C	28	P. S.
SND-30	Man in the Sea	B	28	J. S.
8-98	Life in the Sea	C	11	E. S
8-200	Plankton and the Open Sea	C	19	S.
McGraw-Hill	Conquering the Sea	C	25	J. S.
	The Deep Frontier	C	25	J. S.
	Survival in the Sea	C	30	J. S.
Thorne				
593	Horse Clam			
518	Starfishes			

Man vs. Nature
Pollution
Environmental Destruction

	Title	Color/B. W.	Time	Level
8-778	Vanishing Prairie	C	15	J. S.
4-34	Our Soil Resources	B	11	J. C.
4-721	Conserving Our Wildlife	C	11	E. J.
12A-362	Silent Spring I	B	27	J. S.
12B-363	Silent Spring II	B	27	J. S.
12A-367	What Are We Doing to Our World I	C	27	J. S.
12B-368	What Are We Doing to Our World II	C	25	J. S.
8-592	Erosion	C	14	E. S.
8-753	Problems of Conservation	C	14	J. S.
McGraw-Hill	The Dam Builders	C	30	J. S.
N. Y. Times	Crisis of Environment	5 strips w/records		
M. L. A.	Man's Impact on Environment	C	18	J. S.

Behavior
(territoriality)
reasoning, imprinting

	Title	Color/B. W.	Time	Level
4-467	Environment - Survival - Trout	C	10	E. J.
4-74	Monarch Butterfly	C	11	P. S.
M. L. A.	Checks and Balances in Nature	C	18	J. S.

Thorne

599	Pipevine Swallow Tail
508	Lano Snail

Evolution
(adaptation, speciation)

	Title	Color/B. W.	Time	Level
4-497	Camouflage in Nature - Pattern	C	11	E. J.
4-498	Camouflage in Nature - Form	C	11	E. J.
4-250	How Nature Protects Animals	B	11	E. J.
4-467	Environment - Survival - Trout	C	10	E. J.
8-307	The Cave Community	C	13	J. S.
8-232	Adaptation of Plants and Animals	B	14	J. S.
12-193	The Desert	B	22	E. S.
8-294	Temperate Deciduous Forest	C	17	E. S.
4-74	Monarch Butterfly	C	11	P. S.
12-284	Natures Half Acre	C	33	E. S.
McGraw-Hill	Survival in the Sea	C	30	J. S.
	The Winners	C	30	J. S.

Ecology
Study Methods

	Title	Color/B. W.	Time	Level
8-418	Water's Edge	C	12	E. S.
B. F. A.	Peace and Voices in the Wilderness	C	9 1/2	J. S.
B. F. A.	Which is My World?	C	9 1/2	J. S.

FREE FILMS

1. "The Pursuit of Cleanliness" 14 1/2 min. Color 16mm
   by soap and detergent assoc.
   Association Films, Inc.
   600 Grand Avenue
   Ridgefield, New Jersey 07657

2. "It's Your Decision--Clean Water" 14 1/2 min Color 16mm
   soap and detergent assoc.
   Association Sterling Films
   5797 New Peachtree Road
   Atlanta, Georgia 30340

3. A. Nature of Sea Water 30 min. Color 16mm
   B. The Restless Sea 60 min. Color 16mm
   C. Venemous Sea Animals 30 min. Color 16mm
   Kathy Busa
   Graphic Arts Dept.
   Woods Hole Oceanographic Institution
   Woods Hole, Massachusetts 002543
   phone 617-548-1400 ext. 260 or 261
   (All films must be insured for $200 in return mail)

4. A. To Clear the Air 16mm Color 22 min.
   B. Air Pollution and You 35mm 46 frames filmstrip
   C. Beware of Ill Winds 35mm 39 frames filmstrip
   D. What's Your Air Pollution I. Q. ? 10 true/false questions
   E. Air Pollution, the Facts Leaflet
   F. Air Pollution Primer Book 104 pages
   G. It's your problem--Air Pollution Booklet 12 pages
   Spaceport Area Tuberculosis and Respiratory Disease Assoc.
   P. O. Box 1236
   Eau Gallie, Florida 32935

5. "Estuarine Heritage" 28 min. Color 16mm
   Audio-Visual Services
   Bureau of Commercial Fisheries
   1815 North Fort Myer Drive
   Arlington, Va. 22209

6. A. The Third Pollution" 23 min. Color 16mm #AM-1404
   B. "What's New in Solid Waste Managements?" 37 min. #M-2049-X
   C. "The Stuff We Throw Away" 22min. #M-2048-X
   D. "Burn, Bury, or What?" 19 min. #M-2098-X
   E. "A Day at the Dump" 15 min. #M-1600-X

E. "A Day at the Dump" 15 min. #M-1600-X
F. "Waster Away" 22 min. #M-1740-X
G. "Sanitary Landfill: One Part Earth to Four Parts Refuse" 24 min. #M-1740-X
H. "The Green Box" 17 min. #M-2097-X
I. "In the Bag" 19 min. #M-2091-X
J. "Recycling" 21 min. #M-2118-X
K. "500 Dumps" 21 min. #M-2119-X
L. "The Realities of Recycling" 37 min. #M-2120-X

Films A-L deal with solid waste disposal
Order - Request Free Loan
National Medical Audio visual Center (Annex)
Station K, Atlanta, Georgia 30324
Attention: Film Order Desk

M. "Beward the Wind" 22 min. #M-1707-X
N. "The Run-Around" 11 min. #M-1774-X
O. "Pollution" 3 min. #M-1529-X (Song and pollution scene, sung by Tom Lehrer)

The above concern air pollution, order from same address as solid waste.

Also: "Pandora's Easy Open Pop Top Box" 15 min. (Rural pollution by urbanization) same as above

"Teamwork on the Potomac" 1963 16mm Sound 29 min.
Interstate Commission on the Potomac River Basin
203 Transportation Building
Washington, D. C. 20006
Borrower pays the return postage. Can fill about 75% of all requests. Book 2 months in advance.

"Waters of Destiny" 16mm Sound 27 min.
U. S. Army Engineer District, Jacksonville
P. O. Box 4970
Jacksonville, Florida 32201
Attention: TLO
Borrower pays the return postage. Book sixty days in advance.

"River with a Problem" 16mm Sound 29 min.
Consulate General of Canada
Suite 2110 International Trade Mart
2 Canal Street
New Orleans, La. 70130

"Pall Over America" 1965 16mm Sound 15 min.
"Sources of Air Pollution, Effects . . . Control" 1962 16mm
Sound 15 min.
"Take a Deep Breath" 1963 16mm Sound 25 min.

Above films from:
National Medical Audiovisual Center
Film Distribution
Chamblee, Georgia 30005
Borrower pays the return postage. Book well in advance.

"The First Mile Up" 16mm Sound 28 min.
Consulate General of Canada
Suite 2110 International Trade Mart
2 Canal Street
New Orleans, La. 70130

APPENDIX D

TEACHER LIABILITY & STUDENT RELEASE FORM

LIABILITY OF TEACHERS

The teacher is not protected by the cloak of governmental immunity. A teacher, like any other citizen, may be held liable for negligent conduct which results in injury to another individual

Most civil cases fall within two categories. The case is either a suit in contract or in tort. A tort is any civil wrong independent of contract. Torts are divided into intentional, unintentional or negligent conduct, and strict liability. Intentional torts include assault, battery, and false imprisonment. Unintentional torts, which are the ones most directly involving the classroom teacher, include those cases in which an individual has failed to maintain an acceptable standard of conduct. That is, he has been negligent or careless, and through this negligence has injured another person.

The Courts have held generally that a teacher owes three basic duties to his class. These duties are:

1. Adequate supervision;

2. Proper instruction;

3. Maintenance of all equipment used in a state of reasonable repair.*

It is the teacher's duty to adequately supervise his pupils at all times. This is especially important in shop classes, laboratory classes, dirver training education courses, and during field trips. Proper pre-instruction is necessary to thoroughly acquaint the students (and other adults) with the rules and guidelines for conduct prior to embarking on Ecological Field Trips. The students must realize that the field study of ecology is not to be considered as a picnic or opportunity for athletic release of energy. It represents a study that must be taken seriously, with deliberate violation of the rules considered as sufficient grounds for denial of field trip privileges for the balance of the school year.

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* Public School Law Cases and Materials, Kern Alexander, Ray Corns, and Walter McCann, West Publishing Co., 1969

FIELD TRIP INFORMATION

EQUIPMENT NEEDED:

Clothing -

- Hiking shoes or boots (water repellant)
- Long trousers (blue jeans)
- Sweater or shirt
- Rain jacket
- Hat or water proof head covering

Equipment -

- Pencil
- Field study manual
- Field collecting bag
- Data log

Desired

- Sun glasses
- Magnifying glass
- Field glasses
- Camera & extra film

FIELD STUDY RULES:

1. Report to your instructor when entering or leaving the field study area.
2. Never wander off by yourself; always stay with another person and within the area designated by the instructor.
3. Always carry and use tools properly to avoid injury.
4. Walk, don't run. Never push anyone.
5. Avoid trampling, cutting, or marking plants.
6. Never dig up plants unless asked to do so by your instructor.
7. Collect only those plant leaves or specimens designated by your instructor.
8. Avoid disturbing or killing small animals such as insects, frogs, and any other life forms.
9. When walking in the woods, be careful where you walk.
10. Due to the field trip hazards no sandals or thongs may be worn.
11. No transistor radios on field trips as you can not hear birds or rattlers.
12. No knives, axes or machetes are to be carried on the field trip.

ECOLOGY FIELD TRIP PERMISSION SLIP

My (son, daughter) will be attending ______________________________
place, event

on ________________________ with the ecology class. I understand that care will be taken to provide for the safety of my youngster. In order to provide for safety and accomplish learning objectives I agree to the following rules of conduct for this field study. The student will:

1. Report to the instructor when entering or leaving the field study area.
2. Never wander off by himself; always stay with another person and within the area designated by the instructor.
3. Always carry and use tools properly to avoid injury.
4. Walk, not run and never push anyone.
5. Avoid trampling, cutting, or marking plants.
6. Never dig up plants unless asked to do so by his instructor.
7. Collect only those plant leaves or specimens as designated by the instructor.
8. Avoid disturbing or killing small animals such as insects, frogs, and any other life forms.
9. Not wear sandals or thongs due to field trip hazards.
10. Not bring transistor radios on field trips as one can not hear birds or rattlers over loud music.
11. Not bring knives, axes, or machetes on the field trip.
12. Agree to forego all future field trips if these regulations are not followed.

____
Student signature

____
Parent signature

____
Date

APPENDIX E

CONVERSION TABLES

TABLE OF CONVERSION FACTORS

(U.S. units to metric units)

Length

1 inch	= 25.4 mm
	= 2.54 cm
1 foot	= 30.48 cm
	= 0.3048 m
1 statute mile	= 1.609 km
1 nautical mile	= 1.853 km

Area

1 sq. in.	= 6.45 cm^2
1 sq. ft.	= 929.03 cm^2
	= 0.0929 m^2

Volume and capacity

1 cubic inch	= 16.39 cc
1 cubic foot	= 28,317 cc
	= 28,317 liters
	= 0.028317 cu. m
1 quart	= 0.946 liter

Weight

1 ounce	= 28.35 gm
1 pound	= 453.6 gm
	= 0.454 kg
1 short ton	= 907.2 kg

Pressure

1 p.s.i.	= 70.3 gm /cm^2
	= 0.0703 kg /cm^2
	= 0.703 meter of fresh water
	= 5.17 cm Hg
1 in. of mercury	= 25.4 mm Hg
	= 34.54 gm /cm^2

TABLE OF CONVERSION FACTORS

(Metric units to U.S. units)

Length

1 cm	= 0.394 in.
1 meter	= 39.37 in.
	= 3.28 ft.
1 kilometer	= 0.621 mi.

Area

1 cm.^{2}	= 0.155 sq. in.
1 m.^{2}	= 10.76 sq. ft.
1 sq km.	= 0.386 sq. mi.

Volume and capacity

1 cc or ml	= 0.061 cu. in.
1 cu. m	= 35.31 cu. ft.
1 liter	= 61.02 cu. in.
	= 0.035 cu. ft.
	= 33.81 fl. oz.
	= 1.057 quarts

Weight

1 gram	= 0.035 oz.
1 kg.	= 35.27 oz.
	= 2.205 lb.

Pressure

1 gm /cm^2	= 0.394 inch of fresh water
1 kg /cm^2	= 14.22 p.s.i.
	= 32.8 feet of fresh water
	= 28.96 inches of mercury
1 cm Hg.	= 0.193 p.s.i.
	= 0.446 foot or fresh water
	= 0.394 inch of mercury
1 cm of fresh water=	0.394 inch of fresh water

TABLE OF CONVERSION FACTORS

(U.S. units to other U.S. units)

Length

1 inch (in.) = 0.083 ft.
1 foot (ft.) = 12 in.
1 yard (yd.) = 36 in.
= 3 ft.
1 fathom = 6 feet
1 statute mile = 5,280 feet
1 nautical mile = 6,080 ft.
= 2,026.7 yd.

Area

1 sq. in. =0.0069 sq. ft.
1 sq. ft. = 144 sq. in.
1 sq.yd. = 1996 sq. in.
= 9 sq. ft.
1 acre = 43,560 sq. ft.
= 0.00156 sq. mi.
1 sq. mile = 640 acres

Volume (cubic measurements)

1 cu. in. = 0.00058 cu. ft.
1 cu. ft. = 1,728 cu. in
= 29.92 quarts
= 7.48 gallons
1 cu. yd. = 27 cu. ft.

Capacity (liquid measure)

1 pint (pt.) = 16 fluid ounces
= 28.88 cu. in.
1 quart (qt.) = 2 pt.
= 57.75 cu. in.
1 gallon (gal.) = 4 qt.
= 231 cu. in.

Weight (avoirdupois)

1 ounce (oz.) = 0.0625 lb.
1 pound (lb.) = 16 oz.
1 short ton = 2,000 lb.

Weights of water

1 quart = 2 pounds (fresh water)
1 cu. ft. = 62.4 lbs. (fresh water)
= 64 lbs. (sea water)

Pressure

1 pound per square inch (p.s.i.) = 2.31 feet of fresh water
= 2.25 feet of sea water
= 0.068 atm.
= 2.036 in. Hg
1 atmosphere (atm.) = 14.696 p.s.i.
= 29.92 in. Hg.
= 33.9 ft. of fresh water
= 33 ft. of sea water
1 foot of sea water = 0.445 p.s.i.
1 inch of mercury (in. Hg) = 0.491 p.s.i.
= 1.133 feet of fresh water
= 13.60 inches of fresh water

TABLE OF CONVERSION FACTORS*

(Metric units to other metric units)

Length

1 millimeter (mm.) = 0.1 cm
= 0.001 m.
1 centimeter (cm) = 10 mm
= 0.01 m
1 decimeter (dm) = 100 mm.
= 10 cm
= 0.1 m
1 meter (m) = 1000 mm
= 100 cm
= 10 dm
= 0.001 km
1 kilometer (km.) = 1000 m

Volume and Capacity

1 cubic centimeter (cc)
or 1 millimeter (ml) = 0.001 liter
1 liter (l.) = 1000.027 cc
= 1000 ml
= 0.001 cu. m (m^3)
1 cubic meter (m^3) = 10001.

General System of Multiples

Multiple	Prefix	Symbol
10^{12}	tera	T
10^9	giga	G
10^6	mega	M
10^3	kilo	k
10^2	hecto	h
10	deka	da
10^{-1}	deci	d
10^{-2}	centi	c
10^{-3}	milli	m
10^{-6}	micro	u
10^{-9}	nano	n
10^{-12}	pico	p

Area

1 sq. cm (cm^2) = 100 mm^2
1 sq. m (m^2) = 10,000 cm^2
1 sq. km (km.^{2}) = 1,000,000 m^2

NOTE: European usage employs a comma where we use a decimal point and a period where we use a comma (in large numbers).

Weight

1 milligram(mg) = 0.001 gm
1 gram (gm) = 1000 mg.
= 0.001 kg
1 kilogram (kg) = 1000 gm

Weights of Fresh Water

1 cc. or 1 ml = 1 gm
1 liter = 1 kilogram

Pressure

1 gram per square centimeter (gm/cm^2) = 0.001 kg/cm^2
= 1 cm of fresh water
1 kilogram per square centimeter (kg/cm^2) = 1000 gm/cm^2
= 10 meters of fresh water
= 9.75 meters of sea water
= 73.56 cm Hg
= 0.968 atm
1 centimeter of mercury (cm Hg) = 13.6 gm/cm^2
= 13.6 cm of fresh water
1 centimeter of fresh water = 1 gm/cm^2
1 atmosphere = 1.033 kg/cm^2
= 760 mm Hg

APPENDIX F

RESOURCE MATERIALS

TEACHER CLASSROOM USE

TITLE	AUTHOR	COMPANY
1. Sourcebook for The Biological Sciences	Morholt, Brandwein, Joseph	Harcourt, Brace and World N. Y.
2. Laboratory Block Microbes Their Growth Nutrition and Interaction BSCS	BSCS	D. C. Heath Lexington, Mass.
3. Field Ecology BSCS Block	Edwin A. Phillips	D. C. Heath & Co. Lexington, Mass.
4. Laboratory Guide For Biology (Teachers Manual also)	Peterson, Green Nusbaumer et al	Silver Burdett Atlanta, Ga. 30805
5. Standard Methods for Examination of Water and Waste Water	N/A	American Public Health Assoc. Inc. New York, 10019
6. Clean Water Applied Biology	H. M. Freeman	Fed. Water Pollution Central Ad. Dept. of Interior
7. Marine Ecology	Hillary Moore	John Wiley and Sons
8. Fresh Water Invertebrates of United States	Rovert W. Pennak	Ronald Press
9. Planning School Environment	Richard Myshak	Environmental Service Center
10. Odum's Ecology	University of Ga.	Holt, Rinehart Winston
11. Field Activities Package Transect Activities I Transect Activities II Habitat Study-Transect Study Population Variation	Environmental Science Center	Environmental Science Center 5400 Glenwood Ave. Minneapolis, Minn. 55422

STUDENT CLASSROOM AND FIELD USE

12.	Animals without Backbones	Buchsbaum	University of Chicago Press
13.	Ecology	Peter Farb	Time Inc. N. Y.
14.	Life of Seashore	Wm. H. Amos	McGraw Hill
	" " Marsh	Wm. A. Niering	"
	" " Pond	Wm. H. Amos	"
15.	Petersons Field Guide Books		Houghton Mifflin
	Butterflies		
	Animal Tracks		
	Ferns		
	Trees and Shrubs		
	Reptiles and Amph.		
	Eastern Birds		
16.	Zim Books	Shuttleworth, Zim	Golden Press
	Seashore		New York
	Pond Life		"
	Shells		"
	Insects		"
	Nonflowering Plants		"
	Spiders		"
	Insects Pests		"
	Animals		"
17.	Native Trees of Florida	West, Erdman	U. of Fla. Press
18.	Florida Wildflowers	Baker, Mary Frances	N/A
19.	Fishes, Amphibians, Reptiles of Florida	Carr and Gain	U. of Fla. Press
20.	Field Book of Seashore Life	Ralph Waldo Miner	Putnam 1950

21.	Minnows & Models Color & Change Stream Profiles Variation Within Species Contour Mapping Succession in a Micro Aquarium Micro-Climate Measuring Techniques Population Growth Tree Watching Vacant Lot Studies Outdoor Activities Collection Mans Habitat - The City	Environmental Science Center	Environmental Science Center 5400 Glenwood Ave. Minneapolis, Minn. 55422
22.	Colorimetric Procedures & Chemicals: for Water & Waste Water Analysis (with calibration for B & L Spec. 20)		Hach Chemical Co. , Box 907 Ames, Iowa 50010

BIBLIOGRAPHY

Benton, Allen, and Werner, William E. Jr.; Manual of Field Biology and Ecology, Burgess Publishing Company, Minneapolis, 1962.

Kurtz, Herman; Florida Dunes and Scrub Vegetation (Bulletin # 23), State of Florida Department of Conservation, Tallahassee, 1942.

Thurber, Walter A. and Kilburn, Robert E.; Exploring Life Science, Allyn and Bacon, Inc., Atlanta, 1970.

OTHER REFERENCES

Biological Sciences Curriculum Study, Rand McNally & Company, Chicago, Ill..

"Field Ecology" A Laboratory Block by the Biological Sciences Curriculum Study; Edwin A. Phillips, D. C. Heath and Company, 1965

"Life in the Soil" A Laboratory Block by the Biological Sciences Curriculum Study; David Pramer. D. C. Heath and Company, 1965.

"Microbes: Their Growth, Nutrition, and Interaction" A Laboratory Block by the Biological Sciences Curriculum Study; Alfred S. Sussman, D. C. Heath and Company, 1965.

The Source Book of Marine Sciences, State Department of Education, Tallahassee, Fla., 1968.

Environmental Science Center Curricular Materials; R. Myshak, Director. Golden Valley, Minnesota.

Hach Chemical Company, Ames, Iowa.

Millipore Incorporated, Bedford, Mass.

SUPPLEMENTARY TEXTS

1. Web of Life	J. H. Storer	Paper	$ .95	New American Library, Inc. 1301 Ave. of the Americas, New York, N. Y. 10019
2. Edge of the Sea	R. L. Carson	Paper	.75	New American Library, Inc.
3. Sea Around Us	R. L. Carson	Paper		New American Library, Inc.
4. Prevalence of People	Marston Bates	Paper	1.65	Scribner
5. Forest & The Sea	Marston Bates	Paper	1.65	Random House
6. Life of Birds	J. Welty	Hard	12.95	Knopf
7. Living Community	Carl Hirsch	Hard	3.75	Viking
8. Natural History of Marine Animals	MacGinite GE & Nettie	Hard	11.00	McGraw Hill 1949
9. Life & Death of Salt Water Marsh	John & Mildred Teal			Audubon/Ballantine Book

APPENDIX G

WATER & SEWAGE ANALYSIS

INTRODUCTION

Water that is free of disease producing organisms (pathogens) and chemical substances deleterious to health is called potable water. Water contaminated with either domestic or industrial wastes is called nonpotable or polluted water. The objectives of primary concern in providing potable water are freedom from harmful microorganisms and freedom from undesirable or harmful chemicals.[1]

As a potential carrier of pathogenic microorganisms, water can endanger health and life.

The pathogens most frequently transmitted through water are those causing infections of the intestional tract - namely, typhoid and paratyphoid fevers, dysentery, and cholera. The causative organisms of these diseases are present in the feces or urine of an infected person and when discharged may gain entrance into a body of water that ultimately serves as a source of drinking water or as in the case of the Indian River and its tributaries, in the oysters and clams that are eaten raw or the rivers that are used for recreational purposes. Two large industries - tourism and oyster growing both being jeopardized.

The assumption may be made that the objective in the routine analysis of water would be to isolate pathogenic organisms. This is not true for the following reasons:

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[1] Microbiology, Michael J. Pelczar, Jr., Roger D. Reid; McGraw-Hill 1965 Second Edition.

1. Pathogens are likely to gain entrance into water sporadically, however they do not survive for long periods of time, consequently they could be missed in a sample submitted to the laboratory.
2. If pathogens are present in very small numbers, they are likely to escape detection by standard laboratory procedures.

It is known that the pathogens that gain entrance into bodies of water arrive there via intestinal discharges. Furthermore, certain species of bacterial, particularly Escherichia coli and related organisms designated as coliforms, are normal inhabitants of the large intestine of man and other animals and are present in feces. Thus, the presence of any of these species in water is evidence of excretal or fecal pollution of human or animal origin. If these organisms are present in water, the way is also open for intestinal pathogens to gain entrance, since they, too, occur in feces.

Because the laboratory examination of pathogens has the disadvantages stated, attention is directed to the demonstration of species of known excretal orgin, particularly organisms of the coliform group.

The advantages of this method are:

1. Escherichia coli of the coliform group is constantly present in the human intestine in large numbers. It is estimated that billions of these organisms (coliforms) are excreted by the average

person in one day.

2. These organisms live longer in water than intestinal pathogens do.
3. Healthy persons would not normally excrete typhoid organisms, but should one develop typhoid fever, the pathogen would appear in the feces. Therefore, the presence of coliforms in the water is regarded as a warning signal that the water is subject to potentially dangerous pollution.

The coliform group of bacteria includes all the aerobic and facultatively anaerobic, gram-negative, nosporulating bacilli that produce acid and gas from the fermentation of lactose.

Strict attention must be given to the following details when water samples are collected for bacteriological analysis.

1. The sample must be collected in a sterile bottle.
2. The sample must be representative of the supply from which it is taken.
3. Contamination of the sample must be avoided during and after sampling.
4. The sample should be tested as promptly as possible after collection.
5. If there is a delay in examination of the sample, it should be stored at a temperature between 0° and 10°C.

The development of a new method for the bacteriological examination of water has developed in recent years. It is referred to as the membrane filter technique and has been adopted as a standard method. In the procedure which follows this method will be outlined. However, it should be noted that the above details must be adhered to for the collection of water for bacteriological analysis in the Millipore filter membrane method.

This Millipore membrane filter technique has several desirable features such as:

1. A large volume of water sample can be examined; organisms from any given volume are deposited on the disk.
2. The membrane may be transferred from one medium to another for purposes of selection or differentiation of organisms.
3. Results can be obtained more rapidly than by conventional standard methods.
4. Quantitative estimations of certain bacterial types e. g., coliforms, can be accomplished when appropriate media are used.

Sewage is a potential carrier of pathogenic organisms and sewage-treatment procedures must be effective, for if untreated sewage or ineffective treated sewage is discharged into a body of water there is danger of polluting and destroying aquatic and marine life, ruining recreational facilities, and creating public health hazards. This is very important as was stated before for the Indian and Banana Rivers have been noted for their marvelous environment, succulent oysters and a mecca for water sports, boating, sailing, and swimming. This is the reason that a research project such as this can be said to be not only scientific in nature as it surely is, but also, a public service to keep a close check on the amount of bacterial pollution, as well as, chemical pollution.

There is no doubt that in order to have our public officials realize what a situation can develop scientific facts must be presented to them in order that proper legislation be enacted to protect this area from the fate of Lake Erie and many of the rivers and streams throughout the world.

BACTERIA COUNT

SITE II	SAMPLE	1/10	1/100	1/1000
10/13	20/tntc	16/tntc	10/100$^+$	3/14
10/22	-/tntc	6/tntc	12/100$^+$	1/10
10/26	20/tntc	16/tntc	10/100$^+$	3/14
11/2	tntc/tntc	8/47	-/1	-/-
11/5	30/tntc	5/50$^+$	1/27	-/5
11/9	-/tntc	5/tntc	1/30	-/6
11/16	1/tntc	15/tntc	18/100$^+$	3/30
11/23	1/tntc	4/50$^+$	2/5	-/-
12/2	7/tntc	7/50	-/1	-/-
12/7	6/tntc	1/2	-/-	-/-
1/4	2/tntc	-/1	-/-	-/-
1/11	-/tntc	30/tntc	20/tntc	1/tntc
2/1	-/tntc	-/tntc	30/300$^+$	7/50$^+$
2/9	-/tntc	-/tntc	-/tntc	11/100$^+$
2/15	-/tntc	50/tntc	6/tntc	5/tntc
2/22	4/tntc	23/tntc	2/50	-/16
3/15	3/tntc	40/tntc	8/40	-/-
3/24	50/tntc	40/tntc	5/100$^+$	-/46
4/5	-/tntc	40/tntc	23/tntc	-/-
4/21	23/tntc	2/2	1/1	-/-

SITE LOCATION AND DESCRIPTION

Three sites have been tested along Turkey Creek which runs through Palm Bay, Florida. The Tillman Canal originates east of the St . John's River marsh just east of a dike on the eastern edge of the marsh and runs eleven miles to Turkey Creek in Palm Bay. The Tillman Canal drainage constitutes the head waters of the St. John's river, which is an important source of fresh water for this section of Florida.

The sites chosen on Turkey Creek were the same sites assigned to Central Catholic High School research group on water pollution during the past several school years.

Site I is located closest to where Tillman Canal enters Turkey Creek. Sites II and III are located at the bridge on U. S. 1 near the point where Turkey Creek enters the Indian River.

Site II (upon which this report is written) is located on the south side of the bridge where U.S. 1 crosses Turkey Creek. Site III is located on the north side of the bridge and it is directly across from Site II.

Sites II and III are located at the point where Turkey Creek empties into the Indian River.

Site II is located on the south side of the creek. At this site the bank which is fairly steep, and consists mostly of weeds, cattails, and small plants. There is a long ditch (concrete) extending from the road bed down to the creek. This conducts rain water from a good distance on top of the hill to the creek where it flows into the creek from a large drainage pipe.

Just east of the site are homes which have boat docks on Turkey Creek and septic tanks. To the west of the site boats of all sizes are docked and serviced. About 1000 yards from the site is a canal affording access to a mobile home park on water front property (Palm Bay Estates). Further east and still on the south side of the creek is another marina (the Port Malabar Marina) where boats are docked and numerous homes are built. These homes have sewers comprising a part of the Port Malabar Sewage disposal unit.

The sampling for Site II was done from below the drainage pipe and close to the bank. Both Sites II and III are located in the same area, but on opposite sides of the creek inasmuch as the flow rate varies on each side of the creek. The direction of flow also varies. Current flow rate and direction could be a factor in the determination of causes of high bacterial count.

PHYSICAL MEASUREMENT

A number of physical measurements are made in the water pollution project. They are as follows:

1. Time of day sample is taken
2. Wind velocity and direction
3. Current flow and direction
4. Depth of water at site collected
5. Air temperature
6. Water temperature
7. Turbidity
8. Rainfall (over the period between collections)

Time of day is a factor which should be considered at Turkey Creek. The time should be fairly close for each collection as there seems to be a noticeable change in conditions of current flow and depth at various times of the day due to tide action affecting the Indian River.

Wind direction is taken by observing the direction in which the foliage bends; wind velocity is estimated usually as light, moderate, or strong.

Current flow is measured by a cork float dropped into the water at arms length or is placed at the end of a cane pole. This cork float has a measured amount of fishing line attached to it. At the bottom of the three corks through which the string passes is a weight or weights heavy enough to submerge the float just below the surface of the water to prevent any effect by the wind. If the arm-length method is used a measured distance is stepped off and meters/min are recorded. If the fishing pole method is used, the pole is placed perpendicular to the stream and as close to the surface as possible, then the string is released and timed for the known distance at the moment it stops.

Depth of water at the site is taken by a dip stick calibrated in cm, meters, or inches and feet.

Water temperature is taken at the site where the water is collected. This too is recorded in Centigrade degrees.

Water collected for chemical analysis is collected in half gallon plastic containers and the turbidity is run on the Hach colorimeter kit on return to the laboratory. Filter #4445 is used. De-ionized water is used to zero in meter, sample water is used and measured in Jackson Turbidity Units (JTU).

The condition of the site is observed as to anything that might affect the chemical or biological analysis of the water. Dead fish, water hyacinths, ducks, floating or decaying vegetation, new construction, or any other condition that is noticeable at the site is recorded.

One of the most important physical measurements is rainfall. A rain gauge should be located at the collection site or as near to it as possible. There are various types of rain gauges from the commercial test tube type to the home made funnel, plastic pipe, clamps and two by two using a dip stick for measuring. Readings should be taken at least daily and preferably at the same time of day.

SITE CONDITIONS

The site had dead plants floating, hyacinths, decaying plants, murky water and traces of oil and debris on collection dates July 2 through July 23. On July 28 there was a film on the water and dead weeds and grass. On checking further, it was found that defoliant spraying 200 yards from the site at Malabar Road and Turkey Creek was in progress.

Located about 1000 yards from the site is Palm Bay Estates, a mobile home water front development with canals leading into Turkey Creek near The Palm Bay Marina.

In the August 5th edition of Today paper an article, "Seven Private Sewer Plants Must Improve or Close Up," listed Palm Bay Estates Utilities serving Palm Bay Estates Trailer park as one of seven plants included in this report. Jim Stevenson, Brevard Sanitary Engineer, stated that Palm Bay Estates have made "no plans to update their plant." These plants, the article continues, do not meet the minimum requirements of 90% organic material removal during treatment. Phillip E. Searcy, Brevard Environmental Health Director, is advocating that the county should serve as its own law enforcement agency to regulate sewer plants.

The rainfall, which is an important factor in water pollution should be taken daily (preferably at the same time).

CHEMICAL MEASUREMENTS AND ANALYSIS

Chemical measurements and analyses are performed by the Hach Kit method.

Water is collected at the site or sites in half gallon plastic containers. Glass is not used as plastic will eliminate absorption of ions by glass bottles.

The following chemical tests were run:

NITROGEN, NITRATE

Cadmium Reduction Method
(Modified Diazotization [1-Naphthylamine-Sulfanilic Acid] Method)
Range 0 - 150 ppm

This method registers both nitrate and nitrite nitrogen. If it is desired to obtain nitrate content only, it is necessary to run a nitrite test also and to subtract the nitrite nitrogen value from the value obtained from this nitrate test.

Procedure

1. Thoroughly rinse a colorimeter bottle and the polethylene stopper with demineralized water three or four times. This is most easily done by filling the bottle about one-half full of demineralized water, stoppering it, and shaking vigorously and then pouring off the rinse water.

2. Measure 24.5 ml sample of demineralized water by filling a 25 ml graduated cylinder to the 24.5 ml mark. Pour the demineralized water into the rinsed colorimeter bottle.

3. Using the smaller pipette, measure 0.25 ml (250 microliters [250 lambda]) of water sample and add it to the bottle.

4. Add the contents of one NitraVer IV Powder Pillow, stopper

the bottle, and shake vigorously for one minute. If nitrate or nitrite is present, a pink color will develop. Allow an additional three minutes for full color development.

5. While the color is developing, fill the other colorimeter bottle with demineralized water, or other clear, colorless water, and place in the light cell. Insert the Nitrate Nitrogen (Cadmium Reduction Method) Meter Scale in the meter and use the 4445 color filter. Press the light switch and adjust the light control for a meter reading of zero ppm.

6. As soon as the four-minute color development time has elapsed, place the colorimeter bottle in the light cell, press the light switch, and read the ppm Nitrogen, which is present as nitrate and/or nitrite.

DISSOLVED OXYGEN

Modified Azide-Winkler Method with Drop Count Titration (using PAO)

Collection of the Sample

Collect the sample in the 60 ml glass stoppered bottle by allowing the sample to overflow for two or three minutes. The water should be run into the sample bottle through a glass or rubber tube extending to the bottom of the bottle. Withdraw the filling tube while the water is flowing, making certain the bottle is filled to overflowing, and carefully stopper the sample bottle.

Procedure

1. Carefully remove the stopper from the glass stoppered bottle and add the contents of one Dissolved Oxygen I Powder Pillow and the contents

of one Dissolved Oxygen II Powder Pillow.

2. Stopper the bottle carefully so that air is not trapped in the bottle. Invert several times to mix. A flocculant precipitate will be formed which, if oxygen is present, will be brownish-orange in color. Allow the sample to stand until the floc has settled and leaves the upper half of the bottle clear. Again, invert several times and let the sample stand until the upper half of the bottle is clear.

3. Carefully remove the stopper and add the contents of one Dissolved Oxygen III Power Pillow. Stopper again and invert to mix. The floc will dissolve and, if oxygen is present, will leave a yellow iodine color.

4. Accurately pipette a 5.8 ml sample of the above solution into the titration flask.

5. Add PAO Solution for Dissolved Oxygen dropwise, counting the drops and swirling the flask after each drop is added, until the sample changes from yellow to colorless. The ppm Dissolved Oxygen is equal to the number of drops added. See Note B.

Notes:

A. To avoid trapping air bubbles in the dissolved oxygen bottle when stoppering, incline the bottle somewhat, and insert the stopper with a quick thrust. This will force the air bubbles out.

B. If increased sensitivity is desired, a 29 ml sample is used. This is measured by carefully pouring off the prepared sample from the glass stoppered bottle until the liquid is level with the mark around the middle of the bottle. The sample is then titrated directly in the bottle. Each drop is now equal to 0.2 ppm Dissolved Oxygen.

C. PAO is not decomposed by bacterial action as is Sodium Thiosulfate, and it is, therefore, inherently much more stable. PAO is, however, decomposed by ultraviolet radiation and should be kept protected from direct sunlight except when in actual use.

pH VALUE

Procedure

1. Accurately measure a 25.0 ml sample of the water to be tested by filling the 25 ml graduated cylinder to the 25.0 ml mark.

2. Add 1.0 ml of Wide Range Indicator. Swirl to mix.

3. Fill a colorimeter bottle with some of the original water sample and place it in the light cell. Insert the Wide Range pH Meter Scale in the meter and use the 4084 color filter. Press the light switch and adjust the light control so that the meter reads at the far right end of the scale.

4. Place the colorimeter bottles containing the prepared sample in the light cell, press the light switch, and read the scale. For colors green, blue, and violet, read from right to left on the upper scale. For colors red, orange, and yellow read from left to right on the lower scale.

PHOSPHATE, ORTHO

Stanna Ver Method
Low Range, 0-2 ppm
High Range, 0-8 ppm
For Water and Wastewater

Procedure

1. Take two water samples by filling two clean 25 ml graduated

cylinders to the 25 ml mark. Pour each into two clean colorimeter bottles. For best accuracy, the temperature of the samples should be 24±1°C (75 F).

2. Add, to each colorimeter bottle, 15 drops of Ammonium Molybdate Solution and swirl to mix.

3. Add the contents of one StannaVer Powder Pillow to one of the colorimeter bottles. Swirl to mix. If orthophosphate is present, a blue color will develop. Allow 10±1 minutes at 24±°C for color development.

4. Place the other colorimeter bottle, which does not contain StannaVer, in light cell. Insert the High or Low Range Phosphate (StannaVer Method) Meter Scale in the meter and use the color filter indicated on the meter scale. Press the light switch and adjust the light control for a meter reading of zero ppm.

5. Place the prepared sample in the light cell, press the light switch, and read the ppm Orthophosphate.

CORRELATION, SPECULATION AND CONCLUSION

The pH determination, the measurement of acid or alkali strength of a solution, has long been recognized as an important part of water analysis. This acid strength varies in pH units from 0 (strongly acid) to 14 (strongly alkaline) with a pH value of 7 expressing neutral solution. The pH of most natural waters falls in the range 4 to 9, but can change significantly due to the addition of industrial wastes.

The amount of phosphate present is one of the most important tests in water pollution because phosphates are widely used in municipal and private water systems, in boiler feed water, in household and industrial detergent formulations, and in fertilizers for agriculture. In order to properly utilize this useful chemical in water systems and boilers, one must know accurately the amount present in the system being treated. There is, moreover, a need to analyze the phosphate in waste streams and in natural bodies of water. A certain amount of phosphate may be beneficial in a natural body of water, but too much phosphate can result in eutrophication, or over-fertilization of the body, with the result that aquatic vegetation grows too rapidly, dies and, in decaying, consumes large amounts of dissolved oxygen from the lake or stream.

APPENDIX H

ECOLOGY PROJECTS

ECOLOGICAL CASE STUDIES

How do we act to correct ecological problems? It is possible to answer this question through studies of specific environmental problems that have occurred here in Brevard County, or elsewhere in Florida. A careful scrutiny of these problems will reveal the political, ecological, economic, and legal aspects which are the basis of environmental decisions. Effective action on behalf of the environment requires knowledge of all of these areas. It is recommended that one or more students should research these topics and report their findings to the class. It might be more meaningful to the students if some of the case studies were taken from Brevard County and/or were currently in the news media. It would be worthwhile to start a vertical file in the classroom covering any environmental problems. The student will learn that individual responsible social action is often an effective way in which to combat ecological problems.

Some examples to help get started are listed here:

A. Cross Florida Barge Canal
(Destruction of the Oklawaha River Forest Ecosystem)

B. Myrex and the fire ants
(The United States Department of Agriculture plans to airdrop poison pellets)

C. The Everglades Jet Port
(A new site is sought)

D. The Pineda Causeway controversy
(Silt destroys the Indian River bottom--workers build an island for nesting terms)

E. Local dredge and fill operations in the Indian and Banana Rivers (State Internal Improvement Board lacks sufficient staff for inspection)

F. Calcite mines in Lake Okeechobee

G. Ecological problems that effect a continued local water supply (The Lake Washington Dam and drainage of the St. John's River watershed)

H. Ecological problems caused by local sewage disposal (Melbourne plant must meet State standards by 1973--effluent is high in BOD and phosphate)

I. Tampa Bay and Canaveral Harbor oil spills (Oil kills marine birds/fish and fouls our beaches)

J. Mosquito control by larvaciding, spraying and impounding (Drain to lower water table--spray is toxic to beneficial insects)

K. Oyster industry (High salinity encourages oyster drills--coliforms cause harvesting to be curtailed)

L. Tourism in Brevard (Plans that might create another Daytona or Miami Beach in Brevard can be debated)

M. Disney Development
Agencies concerned with Environmental Problems:

(1) U.S. Army Corps of Engineers
Jacksonville, Florida

(2) Florida Forest Service
Orlando District
(Free trees are available)

(3) Florida State Board of Health
Brevard County
Rockledge, Florida

(4) State Road Department
Tallahassee, Florida

(5) Brevard County Commission
(See listings in the telephone directory)

(6) City Commissions
(Various towns in Brevard County)

(7) Dr. Maurice Provost
State Department of Health
Entomological Research Station
Vero Beach, Florida

(8) Mr. James T. Floyd, Chief
Information Education Division
Game and Freshwater Fish Commission

(9) Mr. A. W. Wren, President
ASPIRE, A Society to Preserve Indian River Ecology
South Highway AIA
Melbourne Beach, Florida

(10) Mr. Allen Cruickshank
Indian River Audubon Society
Rockledge, Florida

(11) Mr. Jerome Carroll
Conservation Officer
Merritt Island National Wildlife Refuge
Titusville, Florida

(12) Mrs. Cherie Down
Marine Biologist
Brevard County Health Department
Rockledge, Florida

(13) Brevard County Mosquito Control
Melbourne Airport

(14) Brevard County Parks & Recreation
(See listings in the telephone directory)

(15) Mr. Karl F. Eichhorn
Florida Defenders of the Environment and Indian River
Audubon Society

(16) Mrs. Archie Carr, Vice President
Florida Defenders of the Environment
P. O. Box 12601
Gainesville, Florida 32601

(17) Pelican Island National Wildlife Refuge
Sebastian, Florida

(18) Mr. Paul Chell
Indialantic Rotary tree-planting project
(Free trees)

(19) Dr. Arthur R. Marshall, Head
University of Miami
Division of Applied Ecology
Coral Gables, Florida

(20) Mr. Joel Kuperberg, Director
State of Florida
Land Management Agency
Tallahassee, Florida

ECOLOGY IMPROVEMENT PROJECT

It is possible to improve the biological and physical environment "right in our own backyard." It is nice to be able to conserve a great forest on extensive wetlands; however, these natural areas are not readily available to most of us and their proper management is complex and expensive. If we are going to learn to manage our environment, we need to learn the principles through the selection of one or more small local sites which have problems relevant to our every day experiences. These sites should be carefully selected; one site will be near or at student's home and the second site near or on school property. Great care should be taken to be certain that the site is not slated for development into a parking lot or new class rooms. Both aquatic and terrestrial communities should be selected. After the site has been selected and its perimeter carefully delineated, the task turns toward a careful study of what physical, biological, and aesthetic conditions exist. Since this is an environmental improvement project we want to carefully document our pregress with before and after studies on the unimproved and improved site.

Before-type studies will include photographic documentation, line transects and quadrat. The methods for these are described elsewhere in the Guide. Photographs should be made in such a manner that they can be exactly duplicated at the end and during the study period. Preliminary studies should be made in such a manner that the environment is left completely undisturbed. No specimens should be collected or removed from the site. Any changes that are made in order to improve the site should be carefully weighed. Ecologists find that when we tamper with ecosystems we often get unexpected and undersirable side effects. For example, when trash is removed from the site important "cover" for animals may be removed. An old board might shelter a garter snake or a bottle in the Indian River might shelter a fish called the goby. If trash at the project site is unsightly but provides "cover" more desirable and aesthetically pleasing cover such as rocks, logs, bird houses, or bushes, should be sought to replace undesirable trash. In any event, the basic principles of ecology should be studied before any attempt is made to remove or add any biological or physical entity from the "mini-ecosystem." Perhaps it may be decided to let it develop on its own.

Questions for Consideration

(1) Draw and label possible food pyramids for the study sites. Identify producers (autotrophs) and consumers (heterotrophs).

(2) Why are insects and insect eating birds decreasing their numbers? Relate the answer to the food pyramid at the study site.

(3) Identify 3 factors from outside the site that might influence the project area.

INVESTIGATION: WATER BODY USAGE SURVEY

BACKGROUND: Knowing year-round usage patterns for a given lake or river completes a picture of the characteristics of the system which would also include data on water quality. Usage patterns reflect human values and behaviors which ultimately determine the fate of a water body. This data can be used to predict the future quality of the lake, to develop guidelines which will assist in planning for best use and to establish criteria for land development around the water body.

This survey can be used alone, or combined with the water quality information to identify problems brought on by attitudes and usage practices. You will want to get answers to questions such as the following:

How do the people in the community use the water body? Is it viewed as a community resource? a nuisance? with indifference?

The best questionnaires are developed by the students themselves, using researchers and aimed at the collection of data for a specific question. In that case, the following questionnaire may serve as a model.

The questionnaires should be distributed to people in each section of town, and in farm, industrial and business districts. Surveyors should record the distribution location of each questionnaire. A good cross section of age groups should be surveyed in each section of town to aid in the evaluation of results.

The questionnaires can be distributed on one day and collected one or two days later. A brief introduction should be planned, explaining the project to the people surveyed.

When the questionnaires are returned, tabulate the data, grouping it for interpretation. Suggestions for analyzing the results are given. The data, however, may suggest other patterns. Following is a sample questionnaire.

RIVER/LAKE USAGE QUESTIONNAIRE

1. How do you use the river/lake area? (check all that apply)

____________ Swimming
____________ Fishing
____________ Excess Water Runoff
____________ Boating
____________ Water for Cooling
____________ Nature Study
____________ Picnicking

__________ Drainage
__________ Playground
__________ Hunting
__________ Irrigation
__________ Water Skiing
__________ Trapping
__________ Watering animals
__________ Do not use the river/lake
__________ Other:

2. How often do you use the river/lake area? (check one)

__________ Never
__________ 1-5 times per year
__________ 6-10 times per year
__________ 11-15 times per year
__________ more than 15 times per year

3. Where do you live? (write street address or area description)

4. Which age group applies to you? (check one)

__________ 1-10
__________ 11-13
__________ 14-18
__________ 19-25
__________ 26-36
__________ 37-50
__________ 51-60
__________ 61-70
__________ 71-80
__________ 81-90

5. Do you think the river/lake is an asset to (city name) (rank by circling a number)

very important ... of no value
5 4 3 2 1

6. Has the river/lake influenced the development of (city)? (rank this opinion by circling a number)

main influence   no influence
5   4   2   1

7. Has the use of land along the river/lake been well planned? (rank this opinion by circling a number)

excellent planning   unplanned
5   4   4   2   1

8. How is the river/lake area useful to (city name)? (check all that apply)

__________ Scenic beauty
__________ Storm water runoff
__________ Water recreation
__________ Transportation
__________ Boating
__________ Used water runoff
__________ Fishing
__________ Irrigation
__________ Swimming
__________ Flood control
__________ Hiking
__________ Industrial cooling
__________ Hunting
__________ Trapping
__________ Picnicking
__________ Nature study
__________ Playground
__________ other: ____________________

9. What problem does the river/lake present (check those you consider problems or rank them by circling a number. 5 is a great problem and 1 no problem)

__________ Flooding - 5 4 3 2 1
__________ Mosquito Production - 5 4 3 2 1
__________ Drowning Hazard - 5 4 3 2 1
__________ Disease Transmission - 5 4 3 2 1
__________ Bad Odors - 5 4 3 2 1
__________ Many Unsightly Areas - 5 4 3 2 1
__________ Poisoning - 5 4 3 2 1
__________ Other: 5 4 3 2 1